Handbook to the Construction and Use of Insect Collection and Rearing Devices

A guide for teachers with suggested
classroom applications

Gregory S. Paulson

Springer

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A guide for teachers with suggested classroom applications

by

Gregory S. Paulson
University of Shippensburg,
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Springer

A C.I.P. Catalogue record for this book is available from the Library of Congress.

ISBN-10 1-4020-2974-8 (HB) Springer Dordrecht, Berlin, Heidelberg, New York
ISBN-13 978-1-4020-2974-8 (HB) Springer Dordrecht, Berlin, Heidelberg, New York
ISBN-10 1-4020-3020-7 (e-book) Springer Dordrecht, Berlin, Heidelberg, New York
ISBN-13 978-1-4020-3020-7 (e-book) Springer Dordrecht, Berlin, Heidelberg, New York

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Published by Springer,
P.O. Box 17, 3300 AA Dordrecht, The Netherlands.

Printed on acid-free paper

springeronline.com

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Printed in the Netherlands.

Dedication and Acknowledgements

I would like to thank my parents, Mr. and Mrs. Neil A. Paulson, Sr., for their support and encouragement through the years. I'd also like to acknowledge the great influence of Dr. Sally L. Paulson and Mr. Neil A. Paulson, Jr., my sister and brother, on my life. Where would I be today without the Bee Club? I’d like to thank numerous colleagues that have given me ideas over the years especially, Dr. Fred Howard who inspired me to pursue this project and think outside the box and special thanks to Ms. Betsy Ray for designing the plankton net in Appendix A, Dr. Tim Maret for designing the aquatic sampling frame shown on page 30, and Dr. Jay Comeaux for designing the cage on page 94. Finally I’d like to thank my wife for her love and good humor through the years.

About the Author

Dr. Gregory S. Paulson's career in entomology has been devoted to the applied side of the science. He is especially interested in developing alternatives to pesticides for insect control. He has over 50 publications including “Insects Did it First”, a non-technical book detailing advancements, such as Velcro and glue that insects developed before humans. He served as a Peace Corps volunteer in Western Samoa in a WHO filariasis research program and studied plant pathology in Hawaii. Most recently he has studied ant population structure and pestiferous insects in orchards. Presently, he is an Associate Professor and Chair of the Department of Biology at Shippensburg University, PA.

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A Guidebook to the Construction and Use of Common Insect Collection and Rearing Devices with Suggested Classroom Applications

Preface

I am often amazed by the ingenuity of educators and scientists with regard to developing cost effective apparatus and methods for carrying out research. This book is a compilation of techniques and devices I have used throughout my career. I do not claim to be the originator of all of these devices. Some of them are widely used and others are adaptations from ideas presented in scientific literature or from similar commercially available products. My sole purpose in writing this book was to collect a variety of ideas together and make them available in one source. This book is organized into 35 units grouped into six chapters. Each unit includes a materials list, instructions for assembly, construction tips, and, if appropriate, suggestions for classroom projects using the devices including suggestions for simple statistical analyses. Appendix B is a review of simple statistics for use in the classroom. Anyone with basic hand tools, a little skill and patience can construct these devices. Most of them require less than 1-hour assembly time and cost very little because they can be made from recycled items. Older students can help construct these items with adult supervision. Photographs are included to help you visualize the completed item.

Introduction

Insects are great classroom study organisms. They are easy to collect and raise and have a fascinating array of life histories. Because they are small and have tremendous reproductive capacity ecological studies of dispersion, predation, parasitism and reproduction can be studied in compressed timeframes and small areas relative to similar studies of larger organisms. Insects are also important bioindicators of the health of ecosystems. In a small space and with very little cost colonies of insects can be raised in classrooms for use in behavioral and physiological studies. The purpose of this book is to explain how to build and use insect collecting and rearing devices and through explanations of the various techniques stimulate educators to explore the study of insects in their classrooms.

Insects are often given little consideration with regard to humane handling practices. Please remember that insects are living creatures and, as such, are entitled to the same treatment as other living creatures. Teachers should remember that students look to them for clues to the proper way of behaving in new situations. When collecting and working in the “field” educators should teach students to respect the environment. Do not collect more organisms than needed, treat all of the organisms you collect with care, and try not to leave signs of your presence in an area by returning rocks, logs, etc. to their original locations. If you must kill animals you’ve collected, do so quickly and humanely. Be aware that in some areas collecting may be forbidden or may require a permit or license. Finally, if you raise organisms in the lab make sure that they are properly housed and have ample amounts of food and water.

CHAPTER 1

CHOOSING MATERIALS FOR PROJECTS

Plastic vs. Glass

When constructing many of the items described in this book you have the choice of using glass or plastic. There are pros and cons for each. Glass is obviously more fragile and produces sharp pieces when it breaks. On the other hand glass is much cheaper to purchase and is often more readily available than plastic sheeting. Glass is much more resistant to scratching and will not cloud and discolor with age. Plastic is easier to work with because it can be readily cut and shaped with normal woodworking tools. Glass is a little trickier to cut and shape for those without glass cutting experience. My recommendation is to stick with plastic especially if younger children will be using the equipment.

Flexible Tubing

There are several types of flexible tubing that can be used for the projects in this book. I think the best overall choice is Tygon or vinyl tubing. Surgical rubber is made of latex and therefore can trigger allergic reactions in some people. Rubber tubing works well but tends to be thick and will crack and dry out with age. Regardless of the type of tubing chosen pay close attention to the wall thickness, if the wall is too thin the tubing will collapse and crimp too easily. I suggest at least 1/16” as the minimum tubing thickness.

Screening and mesh size.

Many of the items you will read about in this book use screening of one type or another. Before you construct these items you need to consider the size of the organisms you’ll be collecting or rearing. In general normal house screening will work well. I prefer fiberglass to aluminum or brass because it is easier to work with and cheaper although it is not as durable. If you are working with small organisms, use fine mesh nylon fabric instead of house screen. It can be purchased at any fabric store. If you are concerned about mesh size you can buy fabric of known mesh size from BioQuip or a craft shop that sells silk screening supplies. You can also measure mesh size with the aid of a microscope. Monofilament mesh will cost a lot more than non-monofilament mesh. The openings in monofilament are more precise in size because there is no fuzz on the fabric. You probably do not need the extra expense of monofilament fabric for the projects outlined in this book. Nylon stockings also make a good substitute.

Always follow the manufacturers handling and safety recommendations when using any glues, solvents or other chemicals.

CHAPTER 2

ACTIVE COLLECTION DEVICES

ASPIRATORS

Aspirators are among the most effective and easiest to use devices for collecting relatively small insects. Since your inhalation power is used to suck unwitting organisms into the device, aspirators should never be used to collect insects from any location where there might be noxious or dangerous fumes nor from carrion or other decomposing matter. There are also some insects that produce defensive secretions that can literally leave a bad taste in one's mouth. Most notable are ants since they produce formic acid and some of the large true bugs (Hemiptera) e.g. stink bugs that produce compounds similar to fingernail polish remover. If you are going to collect nasty insects or from questionable areas purchase an aspirator that creates suction by blowing through a tube rather than sucking. Be careful to keep your aspirator dry. They do not perform well once they've gotten damp and are difficult to dry out.

There are several types of aspirators. The first type (Figure 1) is simply a rigid plastic or glass tube (about 6" - 8" long, 1/4" - 3/8" inside diameter) inserted into a flexible plastic, rubber or surgical latex tube (about 24" long with an inside diameter large enough for the rigid tube to fit inside). A small piece of fine mesh fabric (a stocking works great) is used to cover the end of the rigid tube before it is inserted into the flexible tubing. This is to prevent you from inhaling insects or insect parts. In the photograph the aspirator on the left is fitted with a disposable pipette (cut the pointed end off) as a mouthpiece. To use an aspirator of this design simply hold the business end of the rigid tube near your prey and inhale sharply. Put your finger over the end of the tube once you have collected your specimen (be careful doing this with stinging insects). A sharp exhalation will expel the critter from the tube into a killing jar, vial, or cage. This is my favorite method of collecting insects. I do not recommend that you use a collection tube with an inside diameter of greater than 1/4" because you may not have the lungpower to use it effectively. The length of the flexible tubing should be no greater than the distance from your armpit to your fingertips. If it is too short you will feel restrained during collection and if it is too long it may crimp and restrict airflow through the tube.

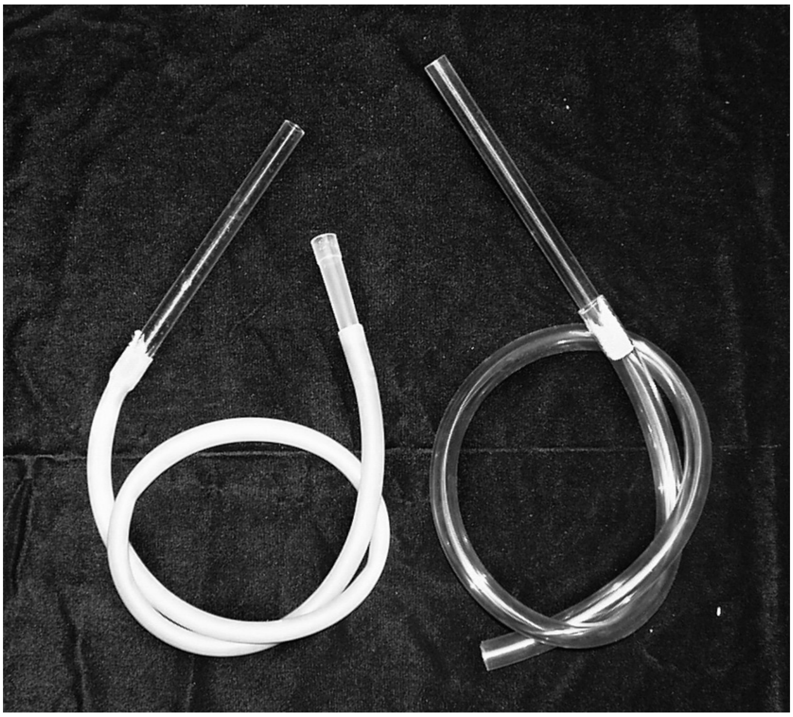


Figure 1. Straight tube aspirators.

The second type of aspirator (Figure 2) uses a collection receptacle to hold organisms as they are collected. The heart of one of these units is a two-hole cork or rubber stopper (#5 or #6 work well). The size of the stopper is dependent on the size of the opening of the collection receptacles you intend to use. You need a good tight fit. You can use a variety of containers. I prefer medicine bottle-type plastic vials with snap caps. In the lower left corner of the photograph you can see an example that utilizes an Erlenmeyer flask. It helps if the collection container is clear and uncolored. You also need two pieces of rigid tubing (1/8" – 1/4" inside diameter, 1 piece - 2 1/2 – 3" long the other about 6" long with a bend in the middle), in my opinion brass works best but aluminum, copper, and glass also work. I don't care for aluminum because it bends too easily. Glass is too fragile. Plastic drinking straws, especially the flexible type (see top center of photo) can also be used but are not as durable.

Cover the end of the short piece of tube with a small piece of fabric, again stocking works well, and with the bottom of the stopper resting on a hard surface push the tube through one hole of the stopper from the top. Stop pushing when the tube is even with the bottom of the stopper. A small amount of lubricant such as saliva, mineral oil or silicone spray will make it easier to push the tube through the stopper. Attach a length (about 24") of flexible tubing to the exposed end of the short tube. Push the second piece of rigid tube completely through the other hole of the stopper so that it projects about 1/4” from the bottom. This small projection helps prevent organisms from escaping the aspirator. This piece of tubing should have a bend in it no greater than 90^0. When bending the material use care not to crimp the tubing.

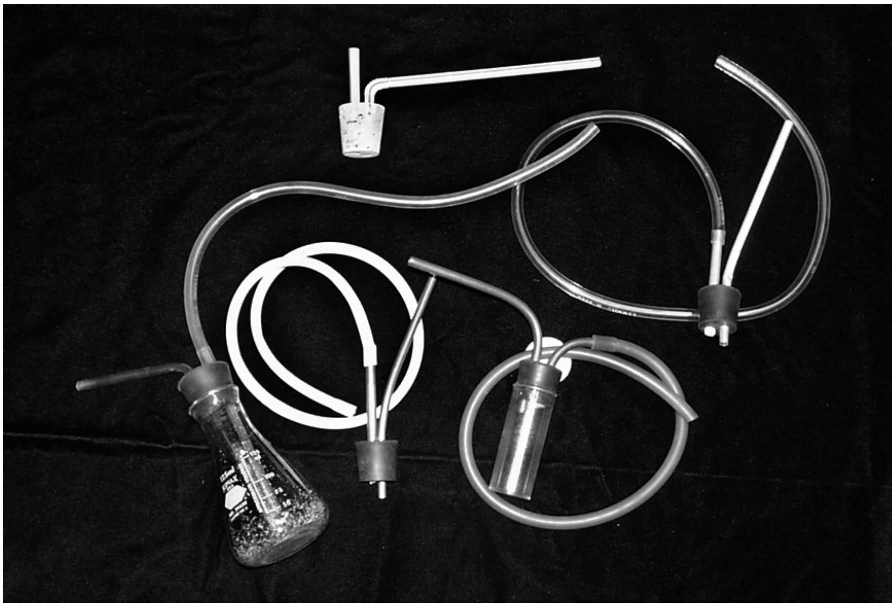


Figure 2. Aspirators fitted with collecting receptacles.

To use your completed aspirator, affix the stopper to a collection container. Point the bent tube at your prey and inhale sharply through the flexible tubing. The critter should be sucked up and into the container. When you have finished collecting, shake the critters to the bottom of the container then quickly remove and cap the container. Put on a new container and continue collecting. This type of aspirator is good if, for some reason (e.g. mark-release-recapture studies), you need to collect a certain number of individuals.

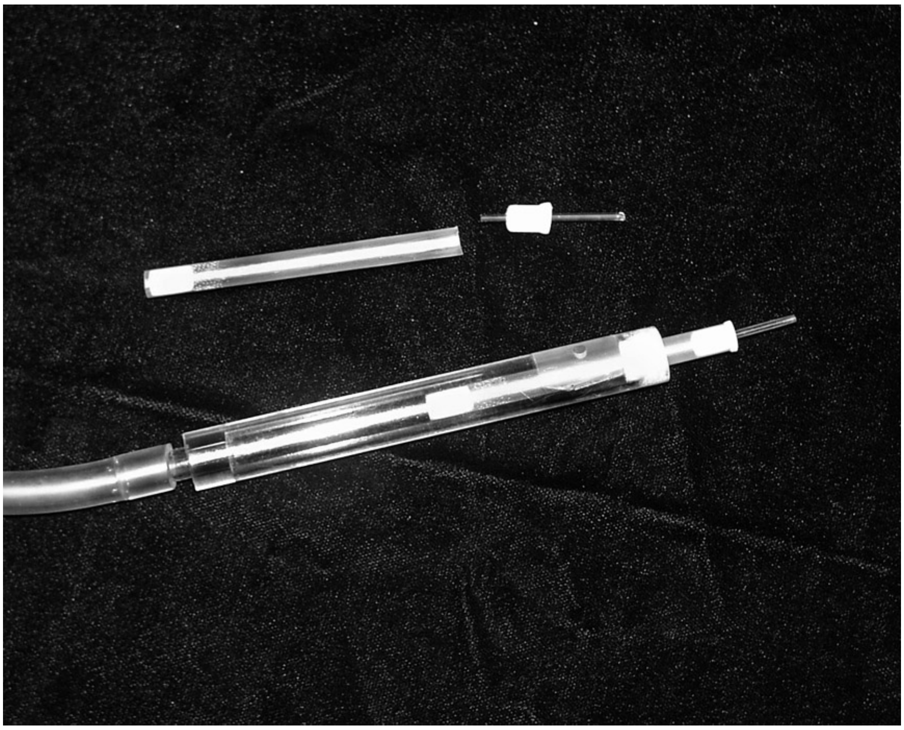


Figure 3. Micro-aspirator for collecting tiny organisms.

If you are working with really small organisms such as thrips (Thysanoptera) or mites (Acari) a mini aspirator is very handy (Figure 3). It features removable collection chambers that allow you to isolate groups of organisms from each other. To construct one of these you will need two different sizes of rigid plastic tube, flexible tubing and self-adhesive foot pads (about 1/16” thick). In the photo the large tube is 4 1/4 “long and has an inside diameter of 3/8”. The large tube must have an inside diameter no smaller than a drinking straw. The small tube is 1” long with an outside diameter of 3/8”. Glue the smaller tube in one end of the large tube making sure to leave about 3/4” of it projecting. After the glue dries attach a 24” long piece of flexible tubing to the projecting end of the small tube. Cut a strip of foot pad about 3/8” wide that is long enough to fit inside of the large tubing without overlapping.

The collection chambers are made from drinking straws, dense foam (1/4” - 1/2” thick) and capillary tubes. Clear straws work best. Cut the straws into pieces about 3” long (the straws in the photo are 1/4” in diameter). Plug one end of each straw with a piece of foam. The foam can be cut with a cork borer. Using a fine needle or insect pin heavily perforate the straw just inside of one of the plugs for about 1/4” (visible in the photograph if you look closely). Take a second foam plug and pierce it through the middle with a dissecting needle and push a piece of capillary tube (about 1 1/2” long) through the hole. The capillary tube should protrude from both sides of the foam about 1/2”. Insert the foam plug and associated capillary tube into the straw.

To use the mini aspirator, insert a straw “chamber” into the holder. At first the fit may be very tight. The coupling will loosen up as the (foot pad) foam is broken in. Leave some of collection chamber protruding from the holder to facilitate removing the chamber. Hold the opening of the capillary tube near your prey and inhale sharply. Exchange collection chambers as needed. When you are using this device it is important to make sure the end of the capillary tube inside of the chamber protrudes beyond the foam plug and does not touch the inside of the straw otherwise organisms may more easily crawl back out of the tube. Organisms can be held in the collection tubes for several days if needed. Capillary tubes with different diameters are available. You can utilize different sizes if you are trying to isolate your organisms by size. Syringe needles can also be used instead of capillary tubes. They are more durable than the glass tubes and also are of known diameter. They can be cut with a hobby or jeweler’s saw, an abrasive wheel on a Dremel-type tool, or a small file. You will want to cut off the sharp point for safety.

HANDLING TINY ORGANISMS

To handle organisms, especially those that are small and slow moving, a pipette tip aspirator is very handy (Figure 4). To construct one of these you will need a piece (8” - 10” long) of small diameter (about 1/4”) surgical tubing and 2 pipette tips (100 microliter work great). Stretch the surgical tubing over the wide end of one of the pipette tips. The other pipette tip is used to make a mouthpiece for your aspirator by cutting the pipette tip in half using a razor blade. Throw away the tip and insert the other piece into the end of your surgical tubing. You can use this aspirator to sort through samples of small organisms like fruit flies or parasitic wasps. Simply touch the tip to the body of the organism while you inhale to pick up and move an individual. The critter should drop off when you stop inhaling. You can also move living organisms like aphids or mites very easily using this device.

Larger pipette tips and tubing can also be used if you are going to be working with larger organisms. You can also attach this device to a small vacuum pump if you prefer not to mouth aspirate. If you try that approach, you will need to drill a 1/8” hole in the side of the pipette tip that you can cover or uncover to control the vacuum.

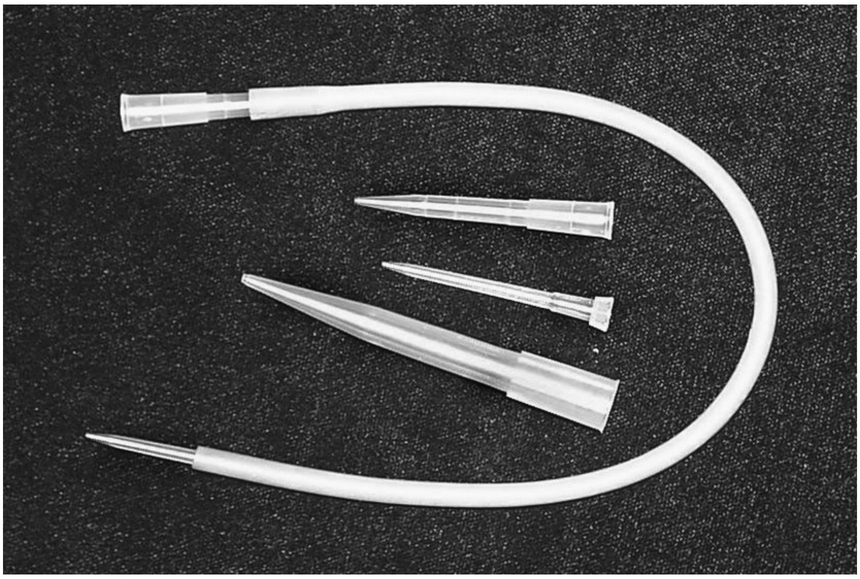


Figure 4. Pipette tip aspirator for handling and moving tiny organisms.

VACUUM COLLECTORS

My graduate advisor, the late Dr. Roger Akre, worked extensively with social insects, as a result we were always collecting nests of ants and wasps. Dr. Akre and his business associate were actually the suppliers of ants for the well-known ant farms that can be purchased in stores. If you want to collect a large number of organisms, a vacuum collector may be a good choice.

For field-work portability is a primary concern so rechargeable and 12 volt DC vacuums are the best choices. There are two major types of portable vacuums: canister-type and “Dustbuster”-type. Canister vacuums do not have a built in power unit so they need to be connected to a power source. Most of them plug into a car lighter or connect directly to a 12v battery. If the latter option is used, be sure to purchase a deep cycle battery (often used for trolling motors). These cost a little more but last longer in the field. You will also want to purchase a battery recharger and perhaps a spare battery. Real Goods Corp. sells wonderful 12v power supplies that plug into any outlet or attach to optional solar panels for recharging. Their performance is comparable to a small deep cycle battery but they are easier to haul around. Dustbuster vacuums have a built in power source that is easy to recharge. Because the batteries are relatively small a Dustbuster will not maintain vacuum power for extended periods of time. I carry as many as six of them in the field if I am planning an intensive study.

Once a sample is taken the contents of the vacuum can be shaken into a large plastic bag for transportation back to the lab. The utility of both types of vacuums can be greatly increased by attaching interchangeable collection chambers to the vacuum. With this modification a vacuum can be used in a manner similar to the aspirators described earlier except you have greater vacuum power. Groups of organisms can be vacuumed up and held in separate containers for further use.

To modify a vacuum you will need about 12” of flexible vinyl tubing (1/2” – 3/4” inside diameter), silicone caulking, super glue or a hot glue gun, rigid tubing (1/2” – 3/4” outside diameter), a small piece of aluminum house screen, and urine sample-sized containers. Cut/melt two holes in each sample container, one in the lid and another near the bottom on one side, with a heated cork borer or razor knife. The hole must tightly fit the rigid tubing. Glue and seal a 2” long piece of rigid tube in each hole. Leave about 3/4” of each tube projecting inside the container. Cut a small circular piece of house screen slightly larger in diameter than the rigid tube. Using a pencil push the screen into the rigid tube in the side of the sample container from the outside until it just protrudes from the end of the tube inside the container. The screen will be somewhat convex when you are finished. The screen will keep your sample in the container. Screen with a smaller mesh can be used if you are planning on collecting smaller organisms but will tend to clog.

The flexible tubing needs to be permanently affixed to the vacuum. This can be easily accomplished by inserting about 6” of the tube into the hose or nozzle of the vacuum, gluing it in place and sealing the rest of the opening with large amounts of caulking. Dustbusters have a baffle just behind the opening of the nozzle that should be removed prior to attaching the flexible tube. The flexible hose attaches to the rigid tube in the side of the sample container; your sample is collected through the rigid tube coming out of the lid.

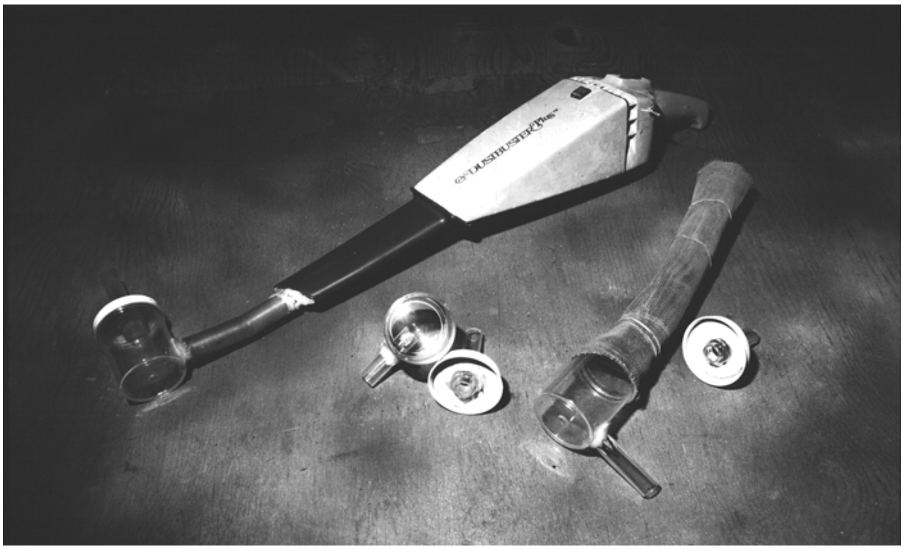


Figure 5. Dustbuster vacuum collector with specimen chambers.

In addition to collecting large numbers of insects a modified vacuum collector is fantastic for conducting mark-release-recapture (MRR) studies. Groups containing known numbers of individuals can be easily captured in each sample container and handled separately. MRR is discussed in more detail in Appendix B.

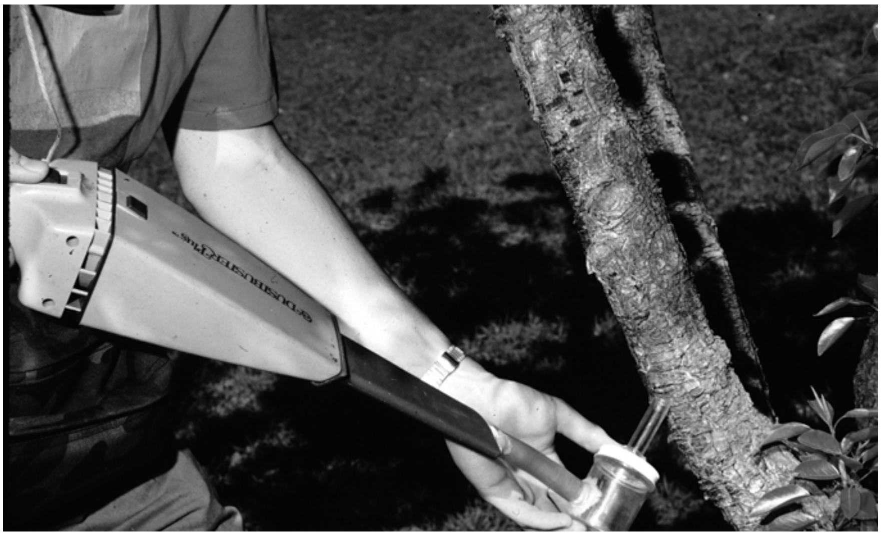


Figure 6. Vacuum collector being used to collect ants from a tree trunk.

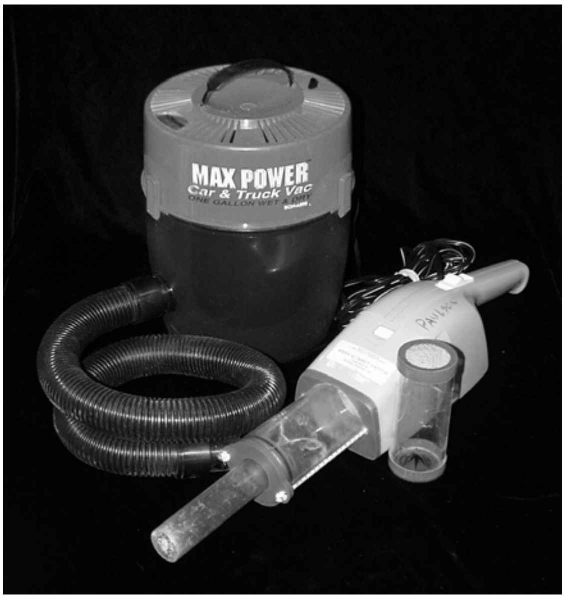


Figure 7. Canister style vacuum collector (left) and commercially available dustbuster style vacuum collector (right)

WHITE PANS

Whether working with aquatic or terrestrial samples a white pan is an extremely useful aid for separating small organisms from soil, vegetation, muck, etc., and should be considered a “must have” for anyone working with invertebrates. When a sample is placed in a white pan it is much easier to differentiate organisms from the matrix of a sample. This is especially true with samples that contain living organisms because their movement against the white background is very pronounced and easy to discern. Just about any type of sample, soil, detritus, algae, even mud, can be examined in a white pan. The most important thing to remember is not to try and “process” too much material at one time. When sorting aquatic samples you can slosh small amounts of water in the pan to help clean up the sample similar to a prospector panning for gold. You can also use a pan as a type of beating tray (page 19) by shaking or jarring a flower or vegetation while holding a pan underneath. Dislodged organisms will land in the tray where they can be easily seen.

Several types of white pans (Figure 8) can be easily purchased. White enamel metal pans are traditionally used but darkroom photography trays are a good alternative. These are much cheaper and lighter to carry since they are made of plastic. Some brands of photography trays are made of fairly rigid and brittle plastic. They are not good choices for field applications but will work well indoors. Enamel pans are very durable but will chip and rust if dented or banged around too much. A good size for a pan is 8” x 12”. I wouldn’t purchase smaller pans unless portability is a major concern.

A durable pan of a good size can be made from the bottom of a 5-gallon bucket (see Figure 8). Most schools have an accumulation of these because they are commonly used for shipping preserved specimens for dissection. You can usually acquire them free from food service (pickles, relish, etc. are shipped in buckets) and construction sites (glues, paint and spackle). At several points around the bucket, measure 4” from the bottom and draw a line. Connect the lines and use them as a cutting guide. A handsaw will do a good job. Use a file or sandpaper to remove burrs and sharp edges. Save the upper part of the bucket since it can be used for a field cage (page 81). A bucket bottom can also be used to protect the bottom of a viewing bucket (page 72).

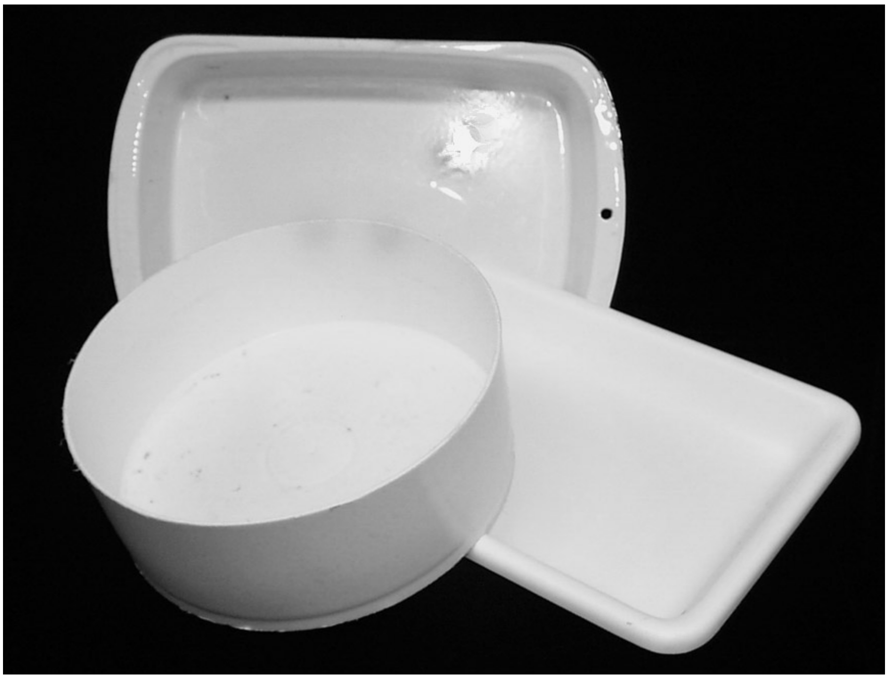


Figure 8. Several types of white pans, tray at front left was made from the bottom of a bucket.

SURBER-TYPE SAMPLER

Surber-type samplers, shown in the photos below, are used to collect organisms from free flowing aquatic habitats. To collect a sample the open frame, which is one foot square, is placed on the streambed with the net trailing downstream. Larger rocks and items enclosed in the sample area are gently "washed" in the mouth of the net. Stream currents carry dislodged organisms into the net. Place the larger items next to the frame as you finish handling them but remember to replace all the material when you are finished. After the larger items have been addressed the smaller gravel and rocks of the stream bottom may be gently disturbed to dislodge additional organisms. Care needs to taken to assure minimal amounts of inorganic material are washed into the net during this step. Contents of the net can be placed directly into containers for later examination or they can be placed into a white pan (page 14) for immediate examination. Each sample is one square foot of streambed.

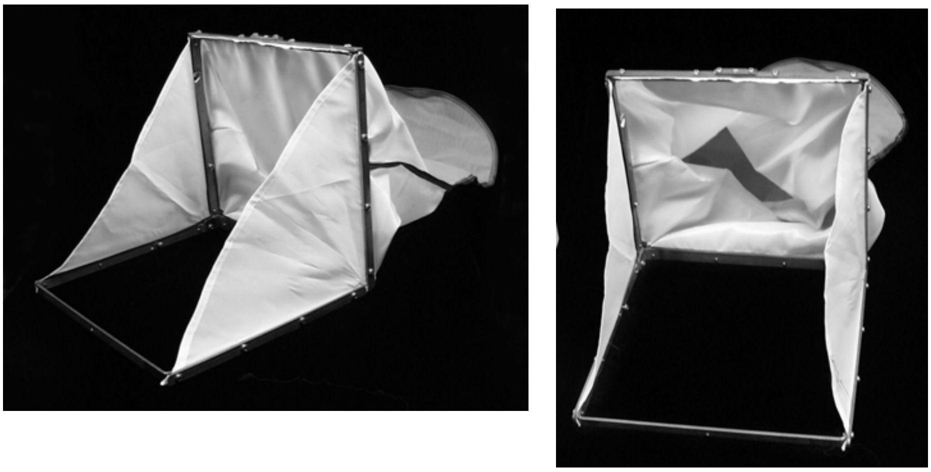


Figure 9. Commercially made Surber sampler.

Surber samplers can be very expensive to purchase but a reasonable substitute can be constructed from a white plastic bucket (see Figure 10, note the net was left off for clarity). The net is constructed from nylon organdy or other mesh fabric. Use the pattern in Appendix A. Using a saw or hot knife cut two rectangular holes on opposite sides of the bucket. Leave at least 1"-2" from the bottom of the bucket for strength. One hole is very large about 1 foot square. This is the water inlet for the sampler. The net attaches at the second hole so it must be sized accordingly. Attach the net to the bucket with 1/8"diam nuts and bolts with washers on the inside and outside to help prevent the net from tearing. When you have finished with these steps cut the bottom out of the bucket. Cutting the bottom out first makes it difficult to cut the openings in the side of the bucket. The sampling area is the open bottom of the bucket. You can easily calculate the sample area using simple geometry.

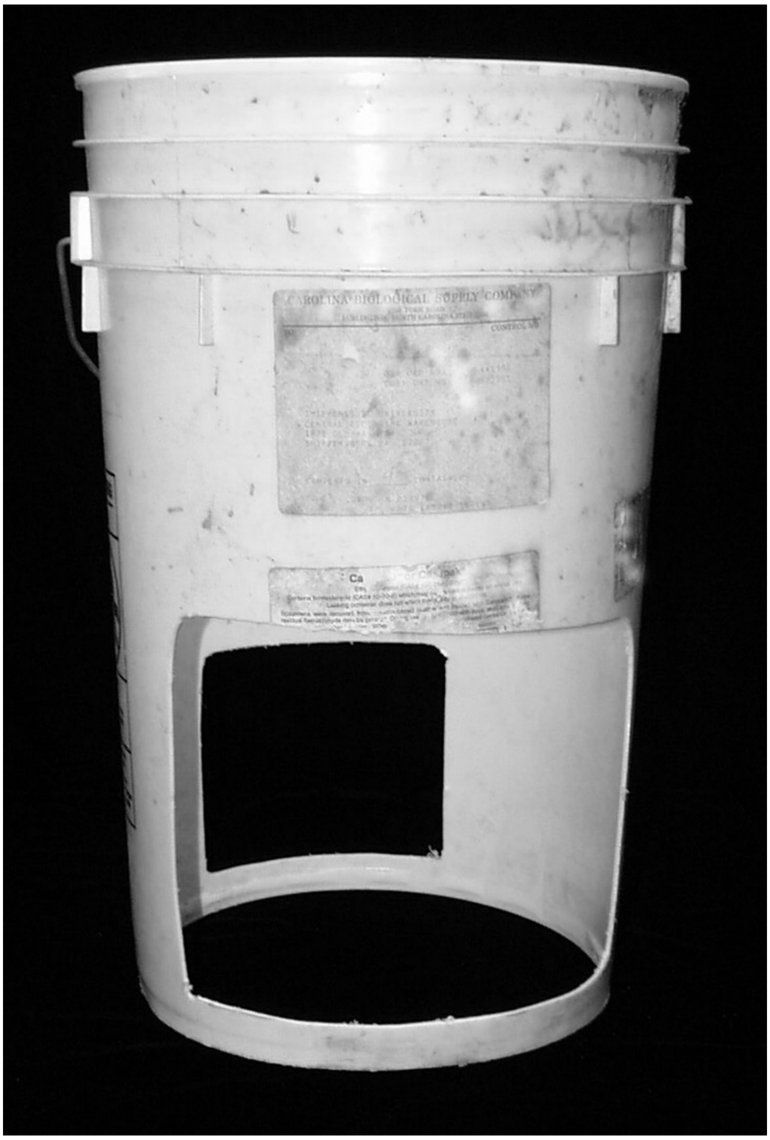


Figure 10. Surber-type sampler constructed from a plastic bucket, note that the net was left out of the photograph for clarity.

There are several simple experiments you can conduct using your sampler. Sample one site throughout the year to observe seasonal changes in population structure, species diversity or growth patterns of individual species. You can also compare samples form different depths or other niches within the stream (e.g. shaded vs. sunny areas, vegetated vs. non-vegetated, riffle vs. pool). If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same sampling effort. Analyze your data using ANOVA or a T-test (Appendix B).

BEATING TRAYS

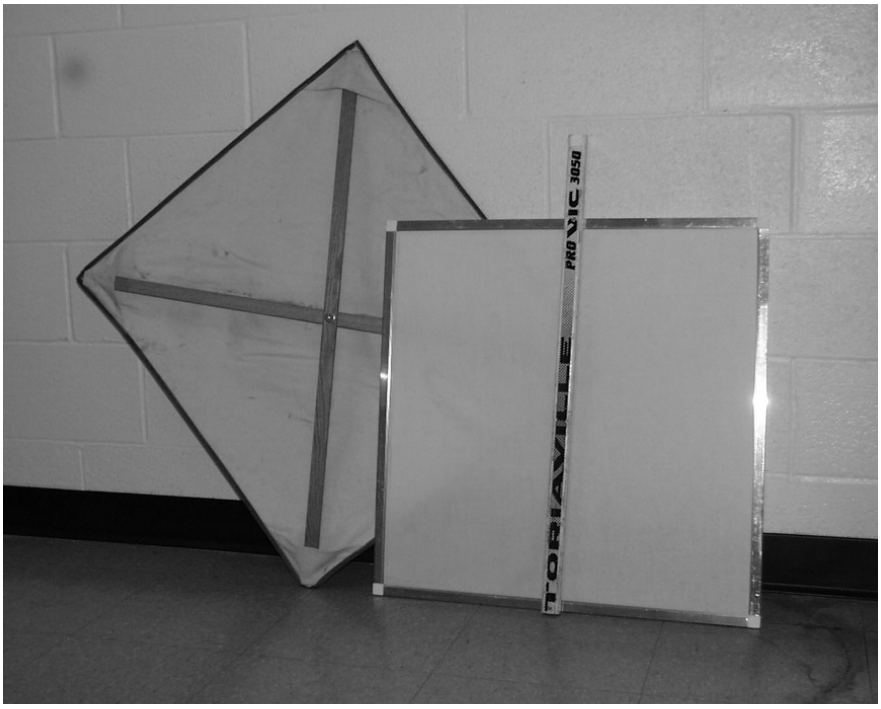


Figure 11. Two types of beating trays, X-brace (left) and aluminum screen frame (right).

Beating trays are another easy way to collect organisms from vegetation. Beating trays are constructed from white or off-white fabric stretched over a frame. There are 2 types of beating trays that can be easily constructed in your home. The first type is constructed from 2 – 36” long pieces of lumber that will form a X-shaped brace over which a square piece of heavy cotton fabric will be stretched. The dimensions of the wood are not critical but should be around 1” X 3/8”. Yardsticks work fairly well especially if each brace is made from two glued together for added strength. Drill a hole in the middle of both braces and use a nut and bolt to hold your braces together. Place a washer on the bolt between the braces to facilitate folding the tray. Use white fabric cut in a square that will fit the frame you constructed. Heavy cotton works well. An old bed sheet will also work but may rip after repeated use. Hem the edges and sew a small pocket across each corner to accommodate the braces (see Figure 11).

The second type of beating tray is constructed from aluminum screen frame and an old sheet. Screen frame is sold in most hardware stores in various colors and sizes. The one shown in figure 11 is constructed from plain aluminum 5/16" frame. Plastic corners and spline (be sure to buy the correct size) are sold separately. Cut the frame with a hacksaw to the desired length. Standard size is 2' square; don't forget to factor in the corners. Once the frame is assembled place a piece of fabric over the frame and, starting in one corner, push the spline into place with the handle of a screwdriver. Use one piece of spline for the entire frame, cut it after you have finished pushing it into place. Thin fabric such as a sheet works well. If the fabric is too heavy you may have difficulty pushing the spline into place. After the spline is entirely in place trim the fabric close to the frame with a razor blade. Next you will need to attach a handle to your frame. There are a lot different materials you can use for a handle. The handle of the beating tray in figure 11 is made from an old hockey stick but a broom handle, electrical conduit, etc. will also work. Attach the handle to the frame with nuts and bolts.

There are several ways to use a beating tray. Hold the tray under a limb and strike the limb sharply with a stick or hose. Be careful to not damage the limb. If you use a stick, you should consider wrapping the end in an old piece of carpet to minimize trauma to the object you strike. You can also shake the limb with similar results. Critters in the foliage will fall on the beating tray where they stand out against the light background cloth. Most of the organisms will be pretty small so keep an aspirator (page 5) handy. You can also take small branches or flower heads and gently slap them against the tray to dislodge critters. Watch out for yellowjacket nests if you are collecting from a tree or shrub. Late in the summer they will be aggressive towards anyone "attacking" their home tree (voice of experience!). If you want to collect from a larger area, use a beating sheet. Spread a bed sheet(s) under a tree or shrub and shake a large number of branches.

You can use a beating tray to monitor small organisms over a period of time (e.g. weekly samples of aphids in trees) or compare beating tray samples from different locations (e.g. species of plants, directional sides of a tree, height in a tree, inner vs. outer foliage, or varieties of apples). Just make sure you use the same sampling effort for each beating tray i.e. shake or jar each limb in a similar fashion. Collect from several sites from each source. Analyze your data using ANOVA or a T-test (Appendix B).

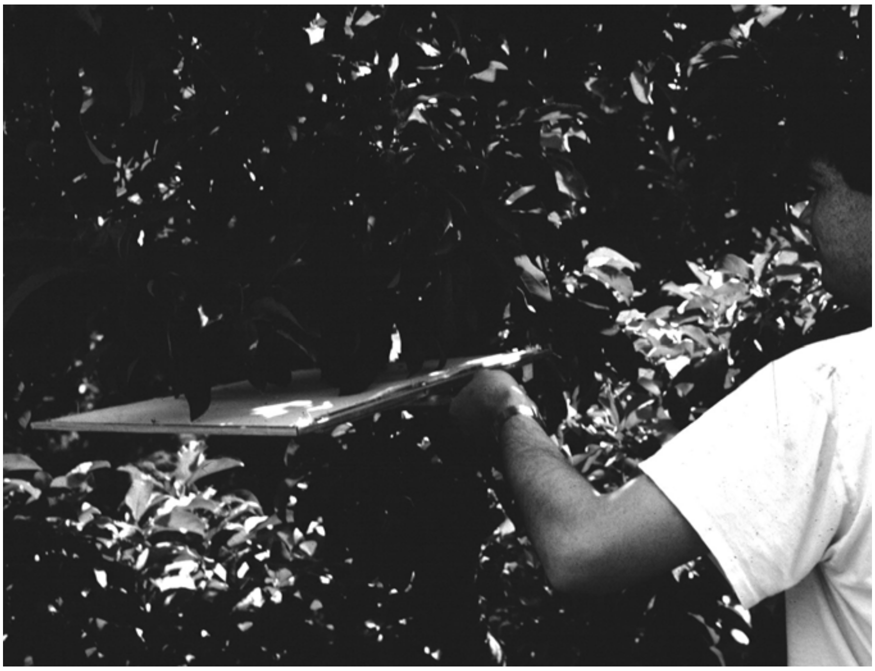


Figure 12. Hold a beating tray under foliage to collect insects that are dislodged.

PLANKTON NET

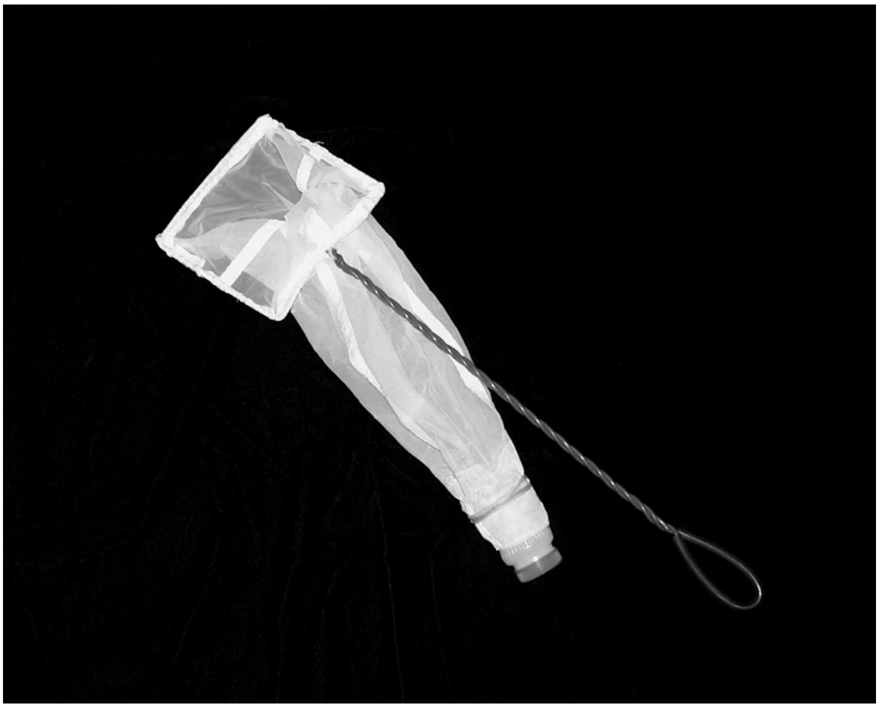


Figure 13. A plankton net constructed from an aquarium net.

If you want to collect very small organisms from an aquatic environment, a plankton net is a good way to go. There are a variety of commercially available plankton nets but they are expensive. A simple and cheap alternative can be constructed from an aquarium net (above). The size of net used is up to you but one with a 5” x 6” opening is a good size. In addition to a fish net you will need nylon organdy fabric (3 nets can be made from 1 yard of 40” wide fabric) and 1” wide twill tape (2 yards) to re-enforce the seams and edge of the net. The net pictured here was made from 2 pieces cut to the shape of the pattern in Appendix A. Remove the mesh fabric from the aquarium net frame and replace it with the net bag you constructed. A juice container (about 10 oz.) was used to form the funnel at the end of the net. Cut the bottom from the container about 4” from the top. Push the container through the end of your net from the inside and glue it in place with durable cement. Model cement and super glue work well. You can leave the cap intact or you can cut away the center portion and glue a bit of mesh over the hole.

To use the net, move it steadily through the water in one direction. If you keep track of how far you move the net you can calculate a rough estimate of the volume of water that went through the net by multiplying the height x width of the net mouth by the distance you moved the net (water movement is a source of error in your calculation). When you finish a sweep of the net you can easily remove the sample from the net by removing the cap from the juice container and washing the net contents into a small jar with a squirt bottle. You can also wash the sample into a white pan if you want to examine the sample more closely. Don't forget to wash the inside of the cap into the jar too. If you will not examine your sample for a number of days, you should store it in a refrigerator or add ethyl alcohol to preserve the organisms in the sample.

There are several simple experiments you can conduct using your net. One pond or site can be sampled throughout the year to observe seasonal changes in population structure, species diversity or growth patterns of individual species. You can also compare samples from different depths or other niches within the body of water (e.g. shaded vs. sunny areas, vegetated vs. non-vegetated). If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same sampling effort. You can also standardize your data by calculating the volume of water. Analyze your data using ANOVA or a T-test (Appendix B).

INSECT NETS

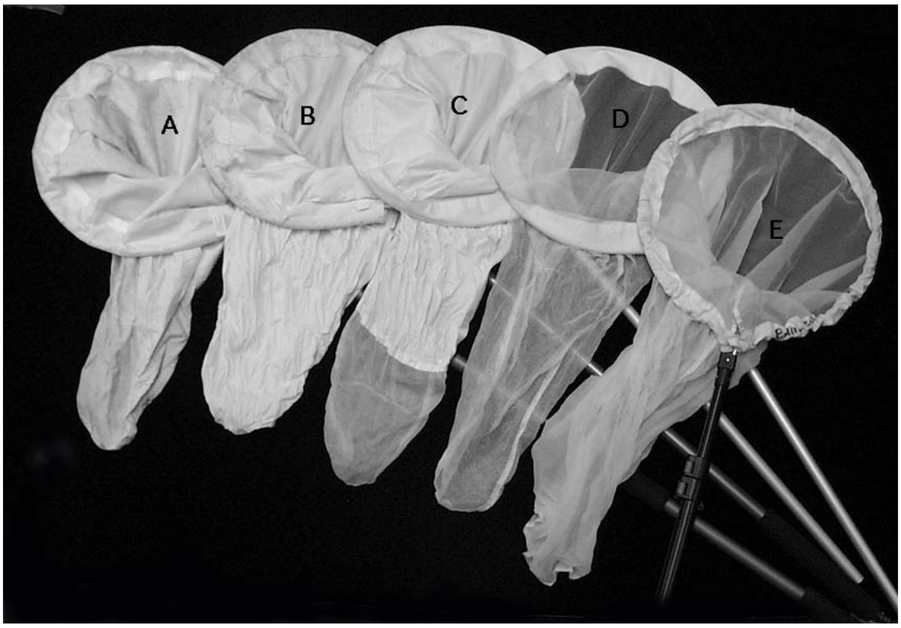


Figure 14. Insect nets: A. Extra heavy-duty sweep net, B. Heavy-duty sweep net, C. Light-duty sweep net, D. standard aerial net, E. Extra fine mesh light weight aerial net.

Insect nets are probably the most misunderstood and misused pieces of collection equipment. Insect nets (above) are constructed from mesh or canvas/cotton twill based on their intended use. It is difficult to make a reliable and sturdy insect net. The weakest part of a homemade net is usually the net ring especially its connection to the handle. Historically a wire coat hanger and an old pillowcase have been the materials of choice for a homemade net but it is difficult to make a sturdy net from that combination and collectors often spend more time fixing their nets than they do collecting. Since nets are relatively inexpensive to purchase I suggest that commercially purchased nets be used.

The aerial net (Figure 14 D & E) is the most readily identifiable type of net. It is the stereotypical “bug net”. As the name would imply an aerial net is often used to capture flying insects although it can be used to collect organisms that are on plants or other surfaces and can be used for very light sweeping (explained below). Aerial nets are usually constructed of nylon or cotton fabric with a relatively tight mesh. The rim of the net is usually made of cotton or canvas to prevent snagging and runs. Very light mesh fabrics (Figure 14 E) are used for collecting delicate organisms such as butterflies. Aerial nets are not intended to be used in water and will be damaged if swept through heavy vegetation. Be especially careful when collecting around burrs, thorns, and other sharp objects.

A sweep net (Figure 14 A, B & C) is designed to make contact with vegetation. The net bag is often partially (Figure 14 C) or completely (Figure 14 A &B) composed of non-mesh material. A sweep net designed for heavy use might even have a rim made of heavy nylon (A in photo) such as what is used in a backpack or tent. When using a sweep net the collector is not identifying a particular organism to capture (i.e. a butterfly on a flower) but rather is sampling the habitat. Sweep net technique is rather simple to learn but does take a little practice to perfect. To explain the technique let's imagine I am interested in using a sweep net to determine the kinds and numbers of organisms that are found in a field of soybeans. As I walk through the field I move my net relatively quickly (to make sure that nothing escapes from the net bag) in a figure eight pattern (Figure 15). I intentionally hit the vegetation with the net bag hard enough to dislodge organisms into the net but not so hard that I wind up with a net full of soybean vegetation. At the end of a sweep I whip the net several times very quickly, again using the figure eight pattern, to drive the critters deep into the net bag and fold the net over unto itself with a final flip. Now you can use an aspirator and forceps to collect insects into vials or killing jars. The entire sample can be dumped into a white pan (page 14) to remove vegetation but flying insects will quickly escape from the sample if you do so. The end of the net can be placed into a killing jar for a few minutes to slow down the organisms before dumping a sample into a white pan.

Sweep netting can be used to sample any type of vegetation but is most effective with herbaceous plants and small shrubs. Be careful around thorns and burrs. Also avoid collecting from wet vegetation. If you are going to compare samples or areas sampled, make sure to use the same sampling effort i.e. do the same number of figure-eight sweeps for each sample and try to walk at the same pace as the samples are taken.

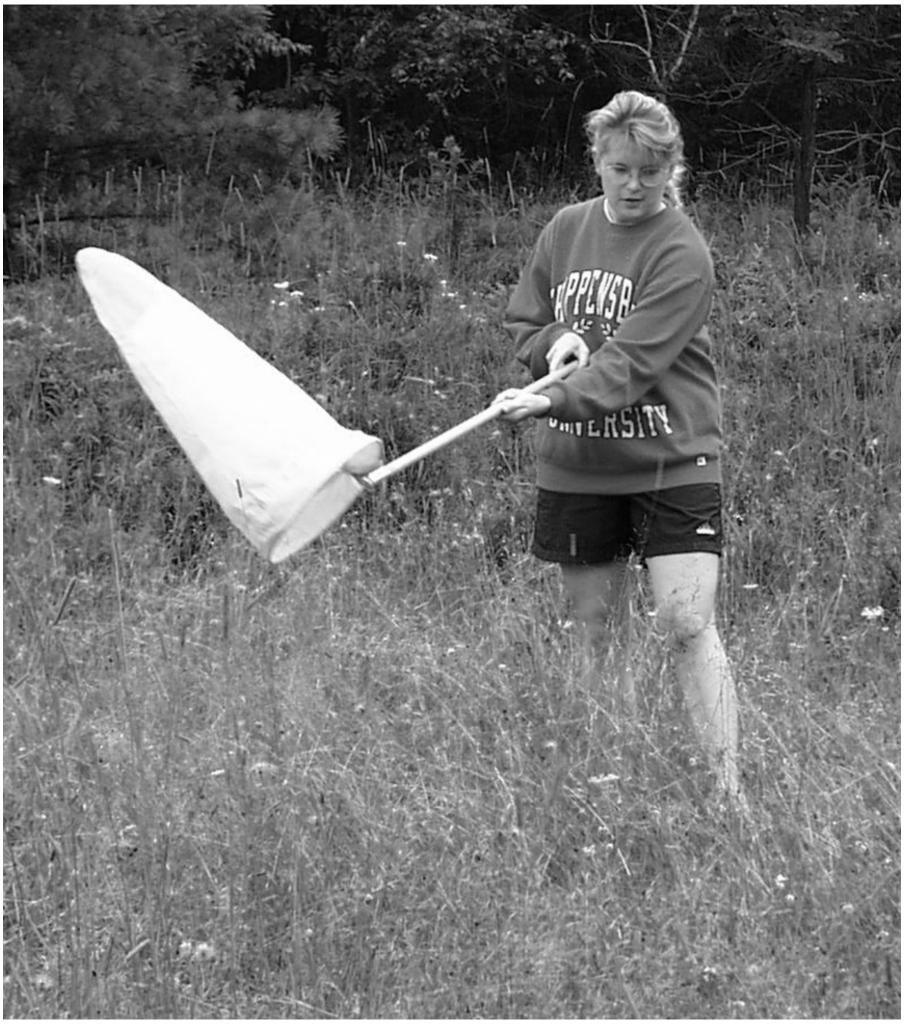


Figure 15. Sweep netting is used to sample a habitat by moving the net quickly in a figure 8 pattern.

Aquatic insect nets are designed to be used in the water. There are several types but the two most commonly encountered are the triangular and D-shaped nets shown in Figure 16. A D-net is generally constructed for heavy duty sampling and they have a heavy metal net ring and double thickness cotton twill sides. Only the back of the net is constructed of mesh fabric. A D-net is used to sample submerged vegetation or sediment. The sample is placed into a white pan (page 14) for sorting after collection. Do not collect too much material at one time as it will complicate sorting the sample and you can damage the net or handle if you try to pick up too much material at one time. Triangular nets are not as heavily constructed as D-nets so they are most suitable for light sampling. A sampling frame (next chapter) can increase the utility of an aquatic net.

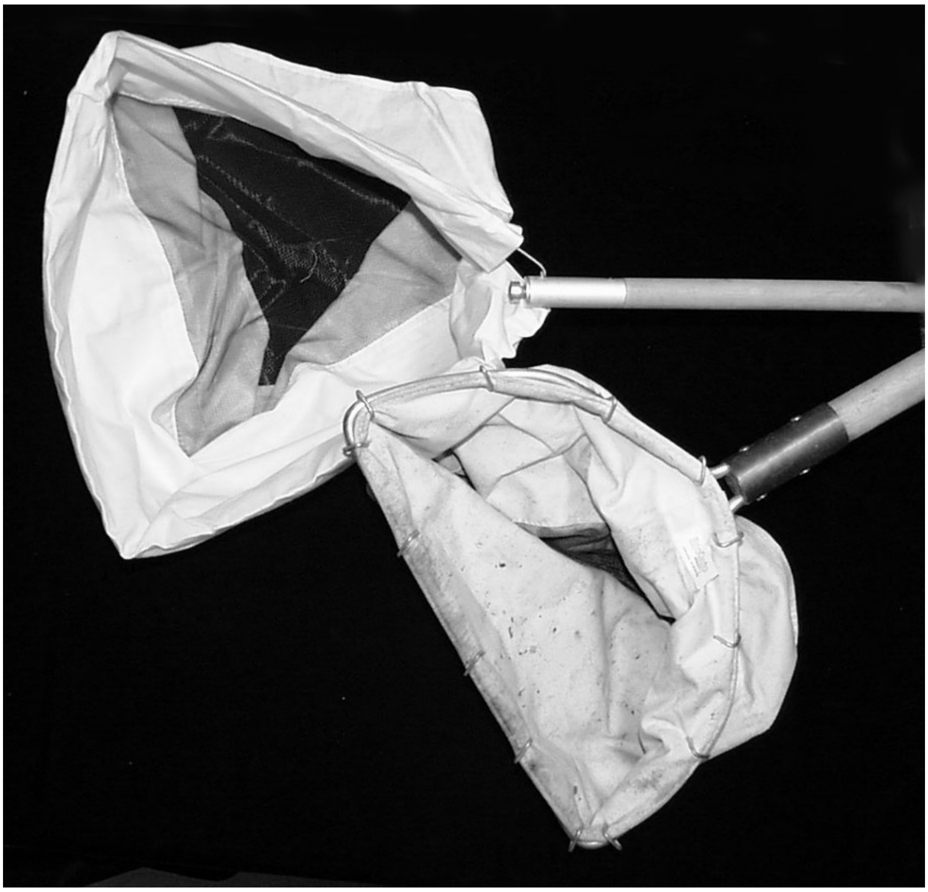


Figure 16. Triangular (left) and D-net (right) aquatic nets.

SAMPLING FRAMES

Sampling frames can be used to help standardize your sampling effort. A terrestrial sampling frame is simply a wooden frame that is laid out on the ground demarcating the area from which you will sample. Everything enclosed inside the frame is considered to be part of the sample. You can place your frame precisely, perhaps by following a predetermined grid pattern, or you can place your frame haphazardly by tossing it in a general direction and sampling wherever it lands. Usually a frame is 1 meter square but other sizes of frames (e.g. 1 yard square, 1 foot square) are

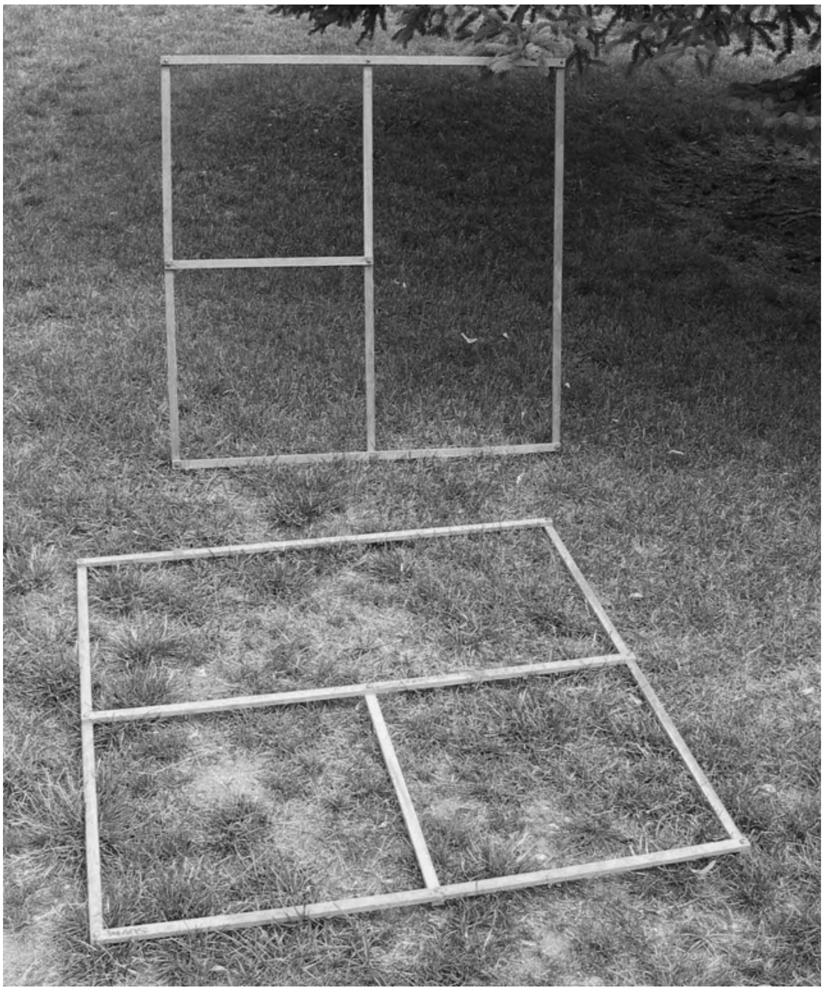


Figure 17. Terrestrial sampling frames with subdivisions.

occasionally used. Some frames will even be subdivided (Figure 17) allowing one frame to have several applications.

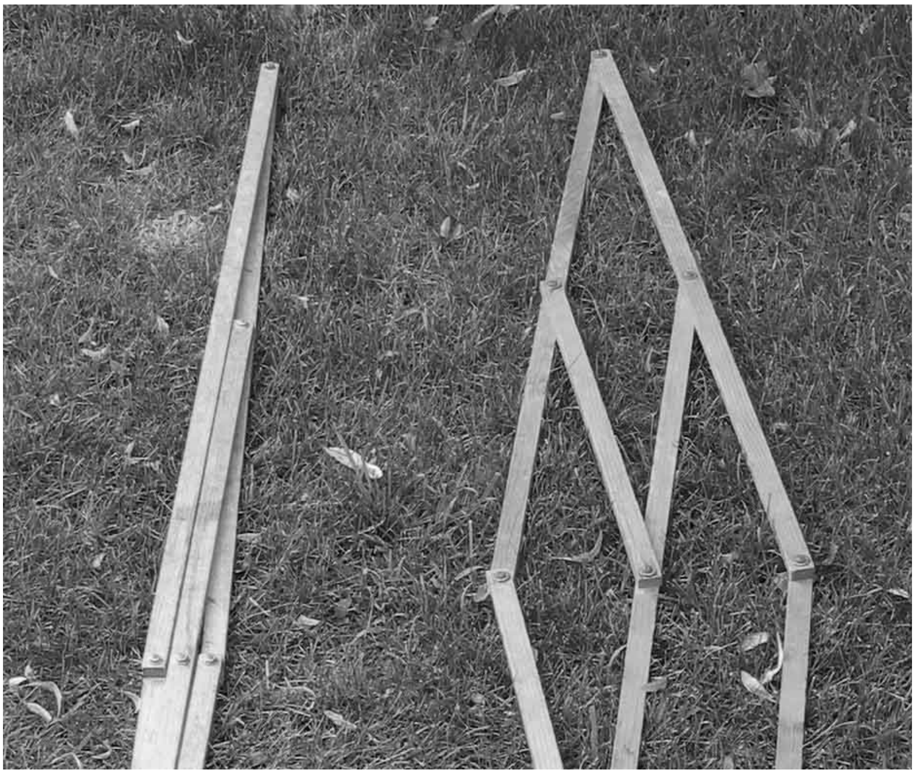


Figure 18. Detail of sampling frame construction illustrating accordion folding action.

To construct a sampling frame for terrestrial habitats you will need 4 pieces of lumber cut to the length that you desire. Thin wood such as furring strips work best but lumber up to 1” x 2” will suffice. Old yardsticks work well for smaller frames. Remember that the **inside** dimension of the frame determines the sampling area. Therefore, the width of the lumber you use must be taken into account in your measurements (e.g. each piece of 1” x 2” lumber cut for a 1 yard square sampling frame must be 3 feet 3 inches in length because a standard 1” x 2” is 1 1/2” wide). Drill a 1/4” hole in each corner of the frame and secure the pieces together with nuts, bolts and washers. Remove the hardware from one corner to fold the frame up for easier transportation or you can fold it up accordion-like.

An aquatic sampling frame is used in a similar fashion as a terrestrial frame, but the high sides prevent organisms from readily leaving the sampling area. It also adds a 3rd dimension to your sample allowing you to sample a volume of water.

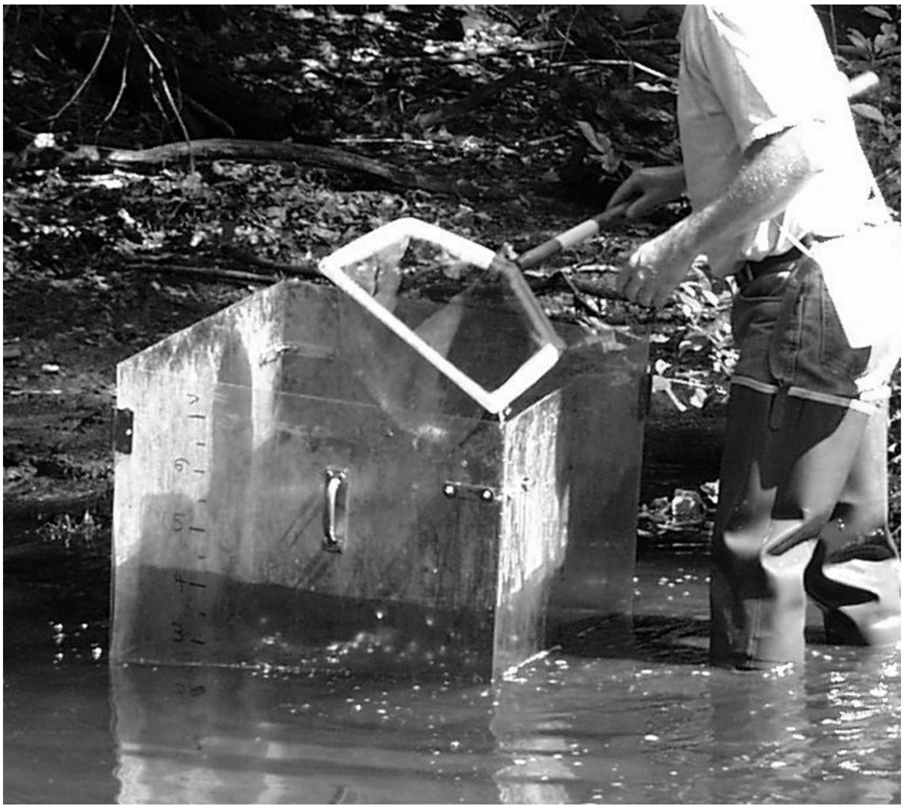


Figure 19. An aquatic sampling frame being used in a pond.

The frame shown in Figure 19 is constructed from 1/4” Plexiglas, 30” high x 36” wide, fitted with hinges on one set of opposite corners and corner brackets on the other set of opposite corners. You will need 4-three inch hinges, 4-four inch corner brackets, and 1/4” diameter x 5/8” long nuts with bolts and washers to construct a frame. The frame in the picture is equipped with handles to facilitate moving it around, but they are not essential. Make sure that the corners are tight when attaching the hardware. For transport and storage remove the corner brackets and fold the hinged sides shut. You can make the sides of your frame higher than 30” but keep in mind that it may be difficult to sample inside a deeper frame. For added strength you may also want to increase the number of hinges and brackets on the corners if you make a larger frame. Mark a ruler on the side of the frame to help you easily determine the depth of the water.

There are several simple experiments you can conduct using your sampling frames. One pond or site can be sampled throughout the year to observe seasonal changes in population structure, species diversity or growth patterns of individual species. You can also compare samples from different niches (e.g. shaded vs. sunny areas, vegetated vs. non-vegetated, different depths of water). If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same sampling effort. Analyze your data using ANOVA or a T-test (Appendix B).

KICK NETS

Kick nets are one of the sampling methods recommended by the U.S. E.P.A (www.epa.gov) for sampling aquatic habitats. They are relatively inexpensive and simple to construct, quick and easy to use, and durable. A standard kick net is 1 meter square. The net portion should be constructed from heavy duty fine mesh nylon netting although fiberglass house screening will also work for most non-technical applications. For rocky-bottom stream sampling, a kick net of 590 micron (#30 mesh size) or 500 micron (#35 mesh size) is recommended. A D-net

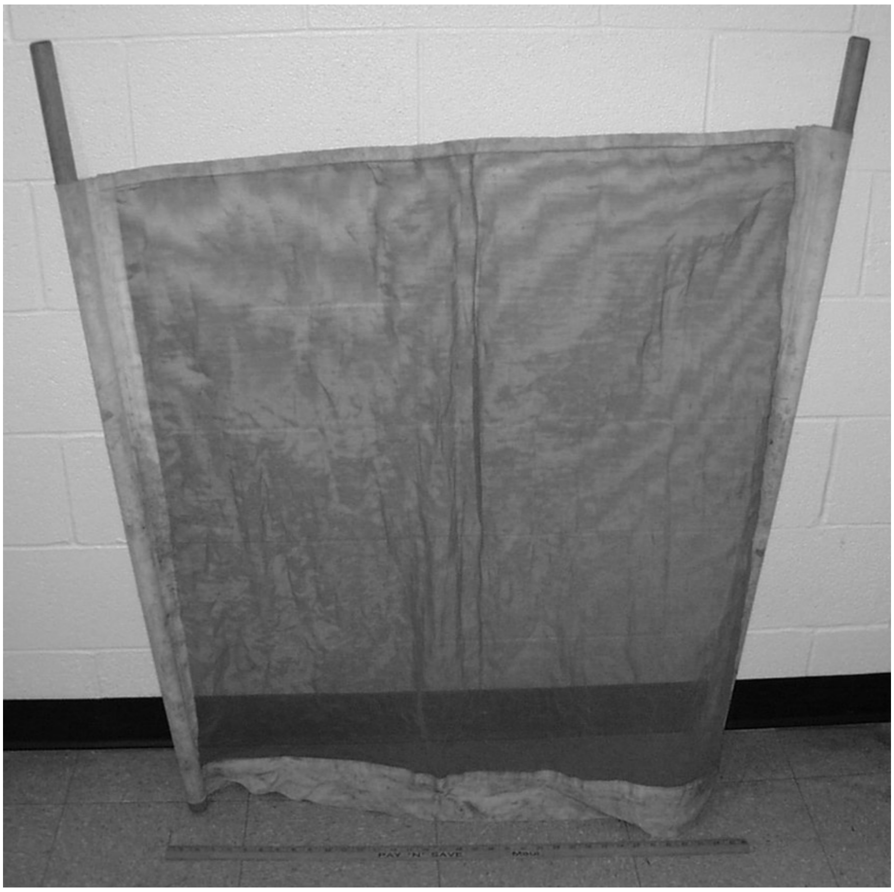


Figure 20. A standard kick net, the ruler at the bottom is one meter long.

(page 24) is better for muddy bottomed areas. Heavy cotton twill or canvas is used to construct pole pockets that are sewn onto both sides of the net. The pole pockets should provide a snug fit for the poles that you have selected. Wooden dowels (minimum 1 1/2" diameter) and handles from old hockey sticks work well. The handles need to project at least 6" from the pole pockets. The top edge of the net should have seaming tape sewn on to help minimize fraying. The bottom edge of the net is constructed from a double layer of canvas 6-8 inches wide.

It is easiest to work in teams of 2 when using a kick net. One person holds the net while the other dislodges the sample. Remember to approach your sample site from downstream so you do not disturb the area before sampling. Stretch the bottom edge of the net along the downstream edge of the area you intend to sample holding it at about a 45^0 downstream angle. Disturb the substrate in front of the net with your feet and hands. Remove the net with a forward scooping action. You can place the sample directly into a container or it can be placed into a white pan for examination in the field.

There are several simple experiments you can conduct using a kick net. Sample one site throughout the year to observe seasonal changes in population structure, species diversity or growth patterns of individual species. You can also compare samples form different depths or other niches within the stream (e.g. shaded vs. sunny areas, vegetated vs. non-vegetated, riffle vs. pool). If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same sampling effort. Analyze your data using ANOVA or a T-test (Appendix B). Visit the EPA web site (www.epa.gov) for specific information about sampling protocols. You can also volunteer to collect samples for the EPA from wetlands in your area.

CHAPTER 3

PASSIVE COLLECTION DEVICES

PITFALL TRAPS

Pitfall traps are probably the most frequently used method of passive collection because they are cost and time effective. They are most commonly used to monitor movement or biodiversity of ground dwelling organism. The basic concept behind the use of pitfall traps is quite simple. A container is buried up to the rim in the soil. Small organisms that stumble into the trap are held until the sample is collected.

Pitfall traps can be made from a variety of containers depending on the purpose of the traps. My personal preference is the 16oz. Solo brand plastic cup. I like these cups because they are pretty tough, and, due to their size, easy to bury even in rocky soil. They are effective for trapping invertebrates yet small enough for most vertebrates that might accidentally stumble into them to escape. Larger traps can be used to monitor vertebrates, I actually have a colleague that uses 5-gallon buckets, but that is beyond the scope of this book. When burying the traps place two nested cups into a hole. Backfill as needed to make the soil level with the cup rim. When that process is completed remove the inner cup. This technique will leave you with a nice clean pitfall trap (Figure 21).

It is essential to use some method of preventing the critters that enter the trap from escaping. In general pitfall traps use a killing fluid in the bottom of the trap to quickly dispatch organisms that fall in. I use environmentally friendly non-toxic antifreeze mixed 1:1 with water. **Do not use regular antifreeze** because **it is poisonous**. Many mammals, especially dogs, find it is very tasty. A 50% alcohol solution will also work, but will evaporate too quickly on hot, dry days. Put about 3/4” of killing fluid in the bottom of each cup. Nesting a small cup from which the bottom has been removed into the trap cup can increase the effectiveness of the traps by preventing larger organisms from climbing out of the trap (Figure 21). If you do not want to use a killing fluid you can coat the inside rim of each trap with “slippery stuff”, a 1:1 mix of petroleum jelly and mineral oil.

When you set-up a pitfall array make sure you make a map to the location of each trap. It’s surprising how difficult they are to find in some habitats. You can also use wire stake flags or other methods to mark the locations but these are not reliable as the sole means of locating traps. Once in place pitfalls should be checked daily to assure that non-target organisms are not harmed. Pitfalls can be used as a short term or long term sampling technique. Here’s a tip: if you plan on maintaining a pitfall array for a long period of time. Use two nested cups in each hole. When you come to remove your sample you can remove the inner cup and replace it with a fresh one. Pour your sample into a small jar for transportation back to the lab. Make sure to clearly label all of your samples. The samples should be carefully examined with the aid of a dissecting microscope or you will miss a lot of small critters. Use a white pan (page 14) to help you sort through each sample. Your samples will contain a large number of ground beetles (Carabidae) and several families of springtails (Collembola). Be on the lookout for the tiniest critters, which may be smaller than the head of a pin. In rainy areas a simple cover constructed from 1/4” plywood with 2” long legs (1” diam. wooden dowel works well) on each corner can be used to prevent pitfalls from filling with water.

You can change the nature of your study by setting up drift fences (Figure 22) in connection with the pitfall traps. A drift fence only needs to be a few inches high and can be easily built from black plastic sheeting. You can buy 50' or 100' rolls of various widths at most large hardware stores. Staple the plastic to wooden stakes to hold it erect. Seal the bottom edge by burying it in the soil. The pitfalls and drift fences can be set up in a "Y" or "X" shape which are designed to direct organisms toward the center of the array or in a straight line. A straight-line drift fence can provide information about directional movement of organisms. Wherever there is a gap in the fence place a pitfall trap.

Ideas for simple studies utilizing pitfall traps:
1. Compare diversity and abundance in different habitats. Around a school you could sample under trees, in a flowerbed, and on the lawn.
2. Paint the inside of the cups different colors and compare the diversity and abundance of critters caught in one habitat but in different colored cups.
3. Do a long-term study of diversity and abundance to monitor seasonal changes in population structure.
4. Use a drift fence in combination with mark-release techniques to study movement from a release point.
5. Compare the diversity and abundance of organisms in one habitat from covered vs. uncovered pitfalls.

If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same effort when examining the sample in the laboratory. Analyze your data using ANOVA or a T-test (Appendix B).

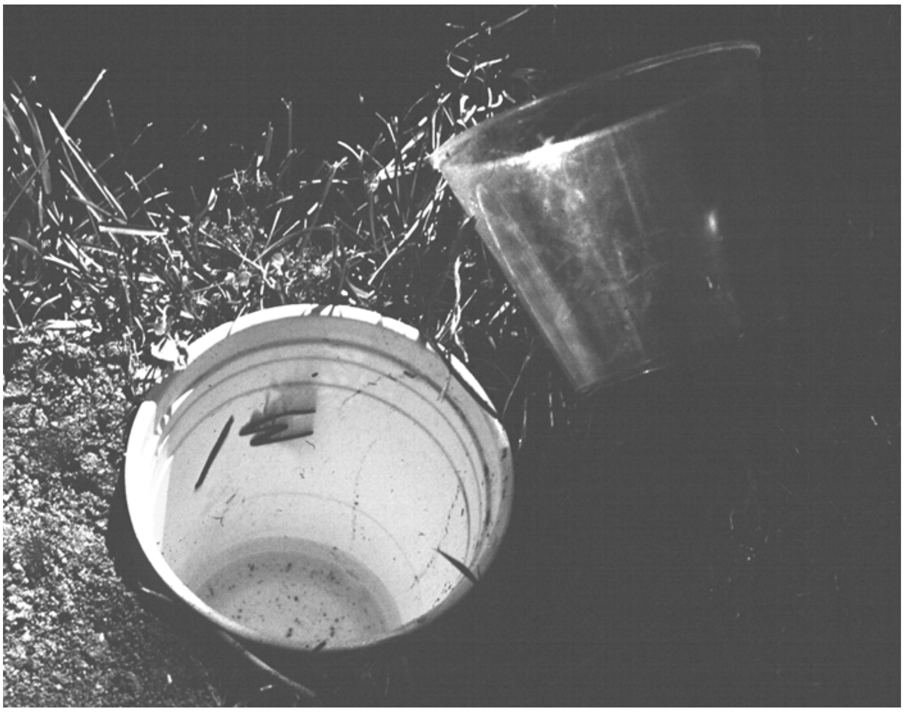


Figure 21. A pitfall trap constructed from a plastic cup, note that only a small amount of fluid is needed in the bottom of the cups. The clear cup at upper right has no bottom and is inserted into the trap to prevent organisms from escaping.

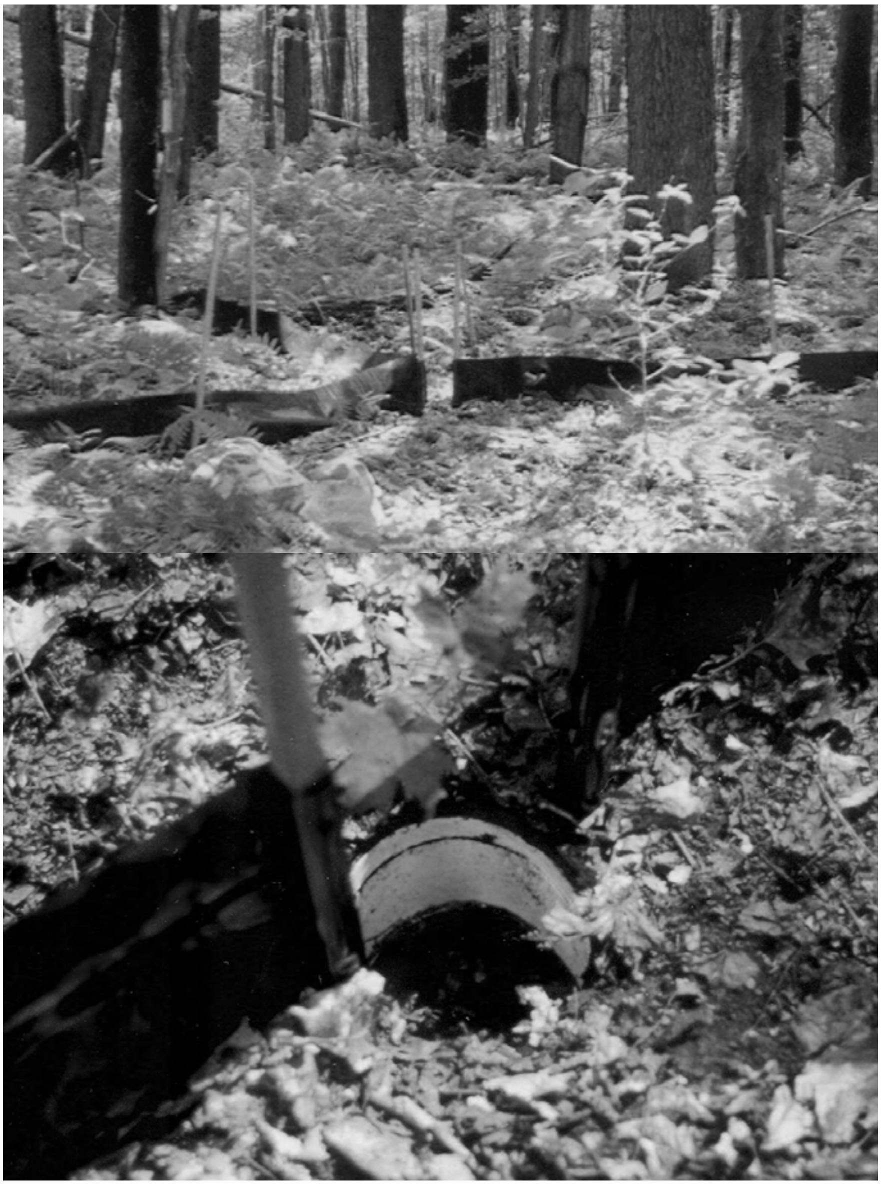


Figure 22. Top, a drift fence in place in a forested area. Bottom detail of the drift fence and pitfall trap.

PAN TRAPS

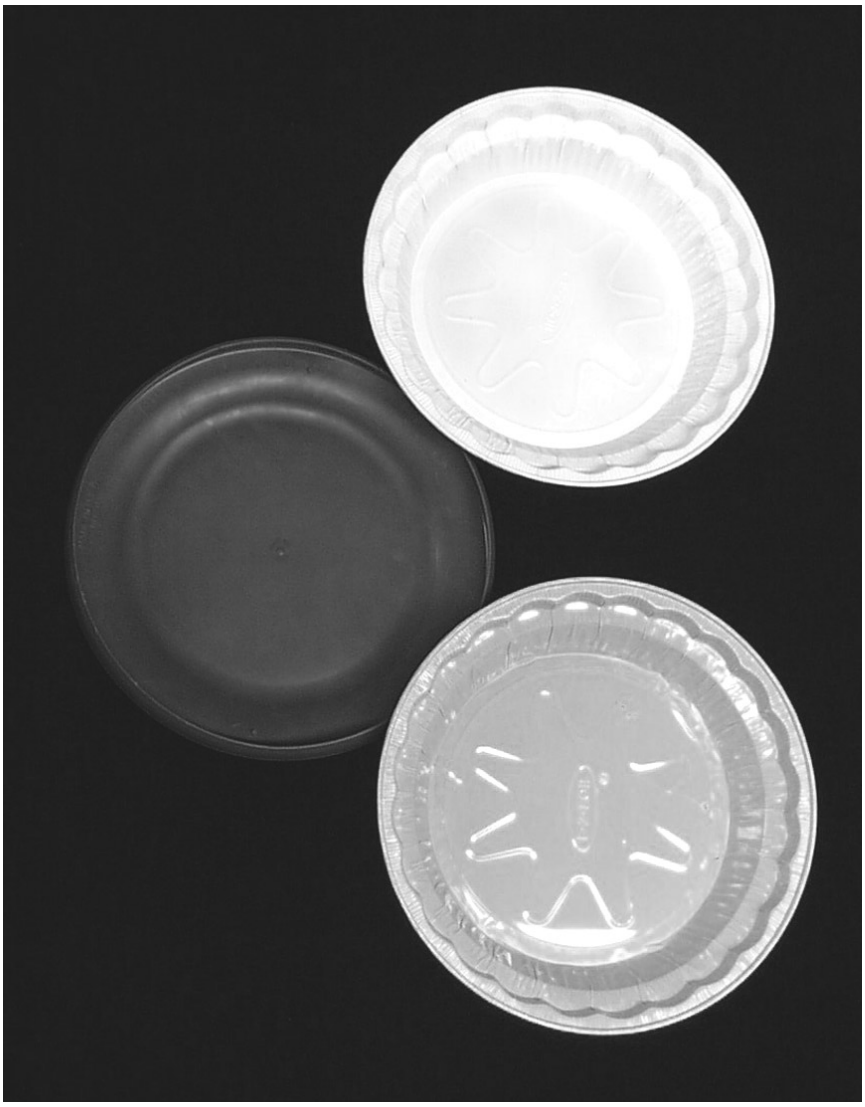


Figure 23. Pan traps made from painted pie tins (upper and lower right) and a flying disc (left).

Pan traps are similar to pitfall traps in some ways but do not target ground dwelling organisms. A perfect pan trap can be made from a disposable aluminum pie pan but other types of pans and even Frisbees (flying discs) will also work. Paint the inside of the pan with a durable paint. White or yellow are good general color choices. Pan color will influence what is attracted to a pan so don't be afraid to experiment. The pans are filled about halfway with killing solution (alcohol or "green" antifreeze) and left out for a pre-determined period of time (usually at least a few days). They should be checked daily to assure that they are intact and non-target organisms are not being harmed. As with pitfall traps make sure you make a map of the pan trap locations. They can be pretty hard to find. In windy areas you may want to put a few stones in each pan to prevent them from blowing away. During collection pour your sample into a small jar for transportation back to the lab. Be sure to label all of your samples. There will not be as much small material in the pan traps as there were in the pitfalls but you should still check each sample closely. A white pan (see page 14) will be very helpful when sorting your samples.

Ideas for simple studies utilizing pan traps:
1. Compare diversity and abundance in different habitats. Around a school you could sample under trees, in a flowerbed, and on the lawn.
2. Paint the inside of the pans different colors and compare the diversity and abundance of critters caught in one habitat but in different colored pans.
3. Do a long-term study of diversity and abundance to monitor seasonal changes in population structure.

If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same effort when examining the sample in the laboratory. Analyze your data using ANOVA or a T-test (Appendix B).

CARDBOARD TRAPS

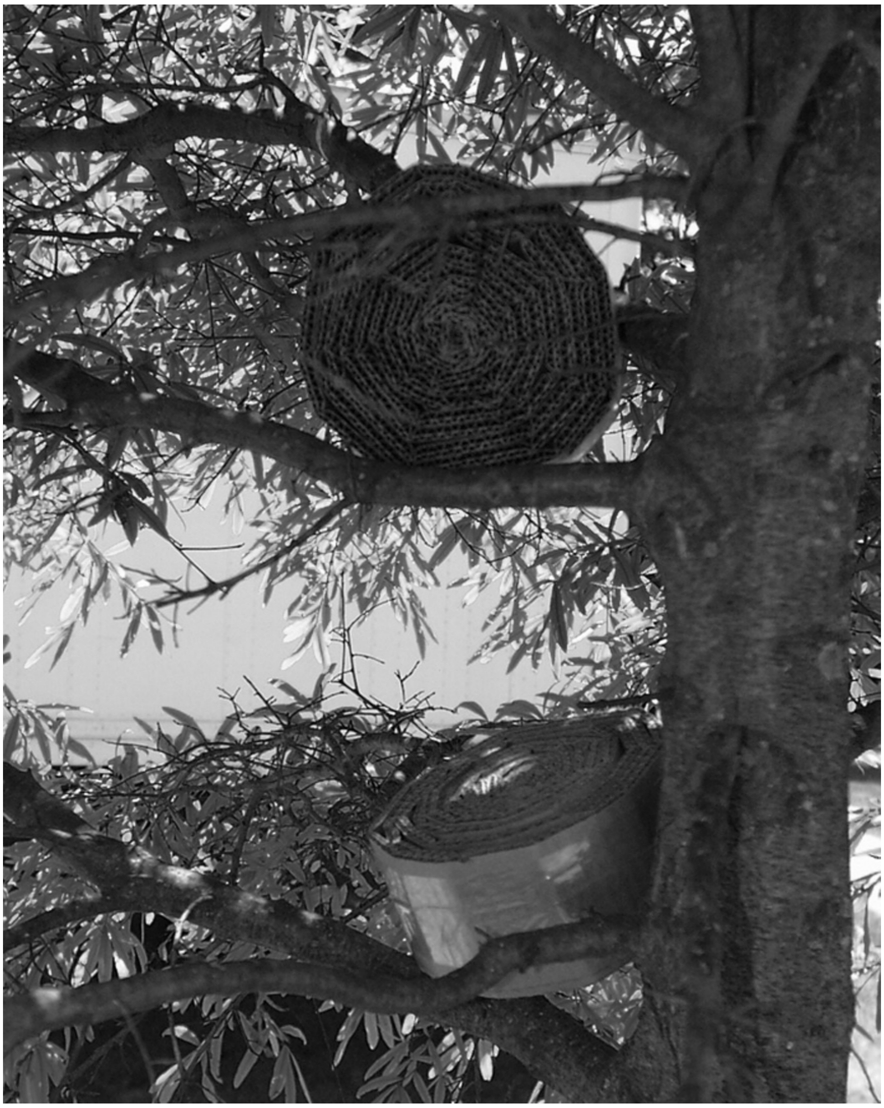


Figure 24. Traps constructed from corrugated cardboard placed in the forks of a tree.

Cardboard traps are good for monitoring organisms moving around in trees or shrubs especially those that are looking for places of refuge to pupate or overwinter. To make a trap cut a piece of corrugated cardboard into a piece 3” – 6” wide and 3’ – 4’ long. The corrugations should be aligned with (parallel to) the short dimension. Roll the cardboard into a tight cylinder; be careful you don't crush the corrugations. Wrap the cylinder with a strong waterproof tape. Go completely around the cylinder with the tape. Once you have finished assembling your traps place them at the study site. You can place them in limb crotches or secure them to the tree with tape or string. You can also place them directly on the ground. The traps can be left out for many months but they will get soggy if there is a lot of precipitation. You can check the traps for the presence of organisms by looking through the corrugations against a light background. To examine the contents of a trap cut the tape, unroll the trap, and peal the layers of cardboard apart. Any eggs, pupae or cocoons you find can be placed in a small cage (page 88) and held until they hatch or an adult emerges, respectively. Don't be surprised if you find parasitic wasps emerging from some of the eggs, cocoons and pupae.

Ideas for simple studies using cardboard traps: in each of these studies try to minimize the variation in the data by making each trap the same size and using the same number of traps for each treatment.

1. Have each student construct several traps and place them at home or around the school ground in the early Fall. Collect them in the early winter and determine what types of organisms are in each trap. Ask the students where they think the organisms would be found in nature at that time of year (probably under tree bark). Discuss the different ways that organisms survive periods of unsuitable weather (hibernation, pupation).
2. Place traps in different parts of the habitat (e.g. limb crotch, limb tip, on ground) and compare the types and numbers of organisms you find in each set of traps.
3. Place traps in different species or varieties of trees but in the same area of each tree (e.g. limb crotch) and compare the types and numbers of organisms you find in each set of traps.
4. Do a long-term study in one habitat. Replace the traps each week. Determine the number and kind of each organism and plot the population density as it changes throughout the year. See if you can predict what will happen in subsequent years.
5. Compare catches from traps with different thickness or corrugation size.

If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same effort when examining the sample in the laboratory. Analyze your data using ANOVA or a T-test (Appendix B).

CARPET TRAPS

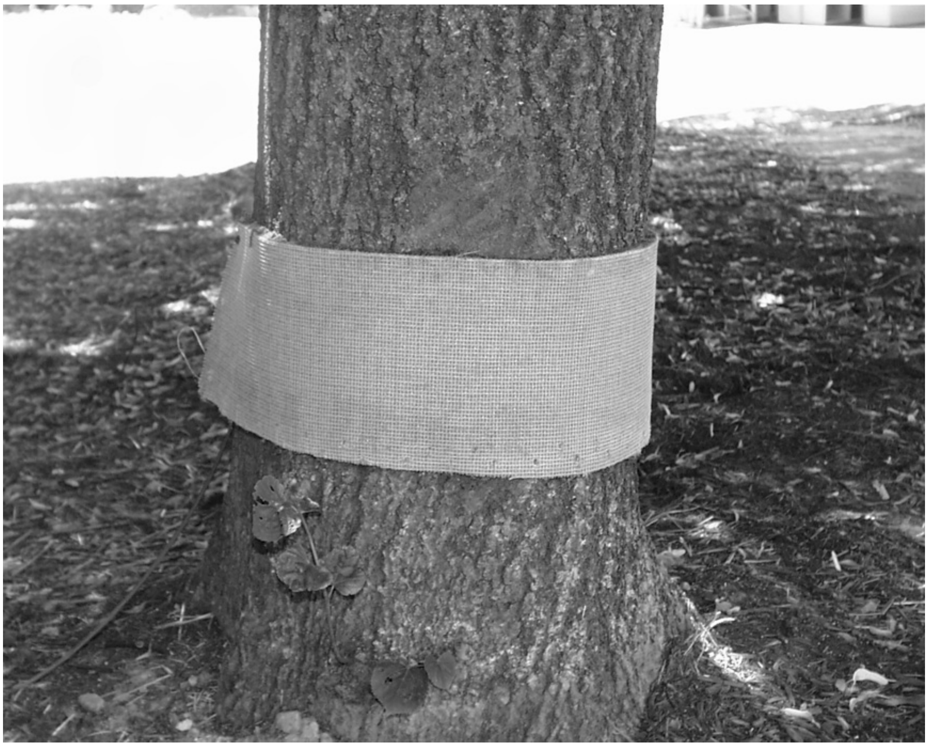


Figure 25. A carpet trap placed around the trunk of a tree.

A carpet trap is a good way to monitor organisms that regularly move around in a tree or shrub. Many foliage eating organisms will move up and down a plant on a regular basis feeding at night and returning to the soil/detritus to escape predation during the day. Carpet traps are an effective way to monitor organisms having that behavior pattern such as armyworms and other caterpillars. Cut an old piece of carpet into a strip about 8” – 10” wide. The length will depend on the size of the tree or branch you are interested in sampling. The piece of carpet should be long enough to go snugly around the tree without overlap. Carpet with a thick or long pile works best. Punch several holes in each end of your strip with a large nail. Wrap the carpet around the tree with the pile facing in. Push heavy cord through the holes you made lacing the carpet strip to the tree similar to lacing up a shoe. The carpet should fit snugly around the tree but you should be able to get 2 fingers under the upper and lower edges without too much trouble. This technique works best on trees that have a relatively rough bark.

You can check the trap each day or leave it in place for a week or more before checking. To check the trap, carefully remove it from the tree and place it pile side up on the ground.

Check the carpet and the tree bark closely for any organisms. It is best if at least two people check each trap that way one person can be stationed on each side of the tree. The larger the tree the more people you should have helping you to minimize escapes and make sure that your data is as accurate as possible.

Ideas for simple studies using carpet traps: in each of these studies try to minimize the variation in the data by making each trap the same size and using the same number of traps for each treatment.

1. Have each student construct several traps and place them at home or around the school ground in the early Fall. Collect them in the early winter and determine what types of organisms are in each trap. Ask the students where they think the organisms would be found in nature at that time of year (probably under tree bark). Discuss the different ways that organisms survive periods of unsuitable weather (hibernation, pupation).
2. Place traps in different parts of the habitat (e.g. main trunk, small limb, near ground) and compare the types and numbers of organisms you find in each set of traps.
3. Place traps in different species or varieties of trees but in the same area of each tree (e.g. 1 foot from ground on main trunk) and compare the types and numbers of organisms you find in each set of traps.
4. Do a long-term study in one habitat. Replace the traps each week. Determine the number and kind of each organism and plot the population density as it changes throughout the year. See if you can predict what will happen in subsequent years.

If you are going to do comparisons, be sure to collect more than one sample on each date or from each site (replications) and be sure to use the same effort when examining the sample in the laboratory. Analyze your data using ANOVA or a T-test (Appendix B).

MALAISE TRAPS

A Malaise trap is designed to collect flying insects. Commercially purchased traps are expensive but are well made and include a collection chamber and poles. It's fairly easy to make an inexpensive one out of nylon or cotton mesh fabric such as would be used in a mosquito net or netting for tent doors. Seam tape should be used to make your trap more durable. White or dark fabric can be used. While there will be a lot of overlap you will catch some different species on a dark vs. light colored trap. Most insects are easier to see on white fabric. The photographs provided here are of a commercially produced net available from Bioquip (see Appendix D).

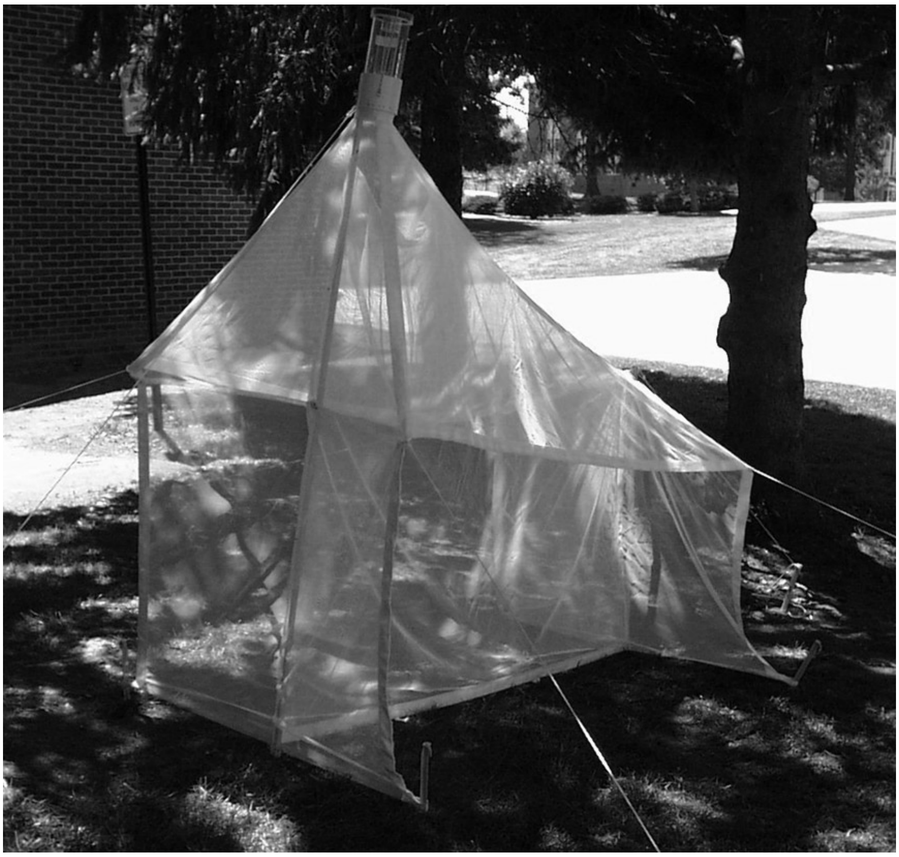


Figure 26. A Malaise trap.

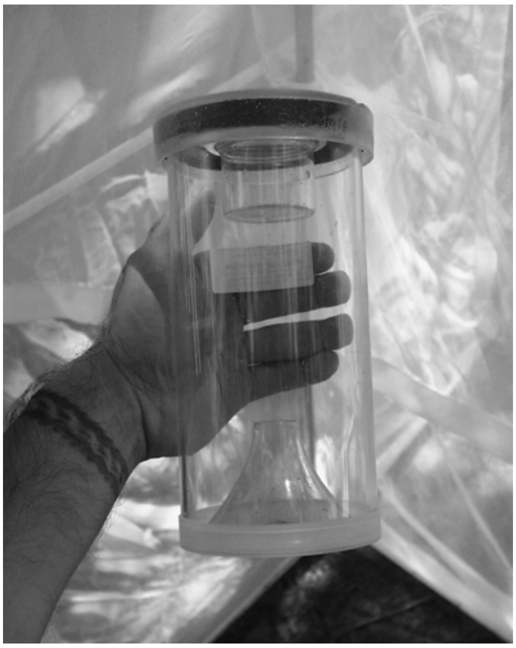


Figure 27. Collection chamber of a Malaise trap, note the inverted funnel in the lower portion intended to trap insects in the chamber.

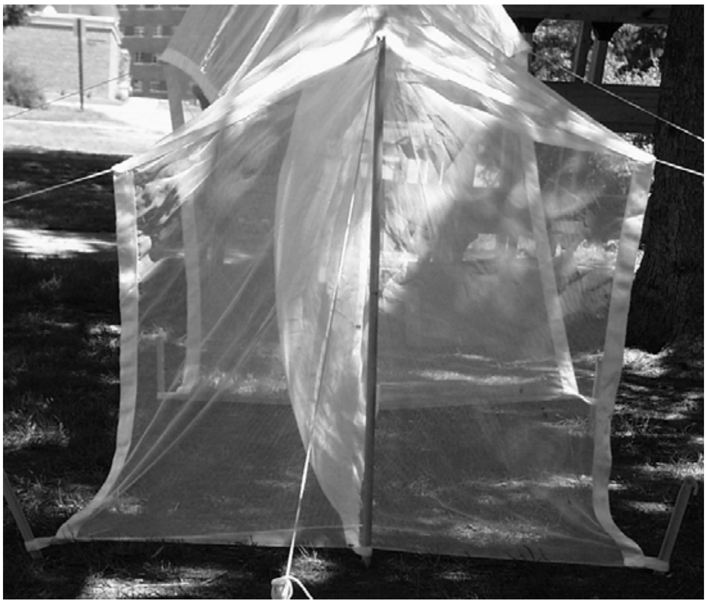


Figure 28. Rear view of Malaise trap; note that the center divider is being blown to the left by the wind.

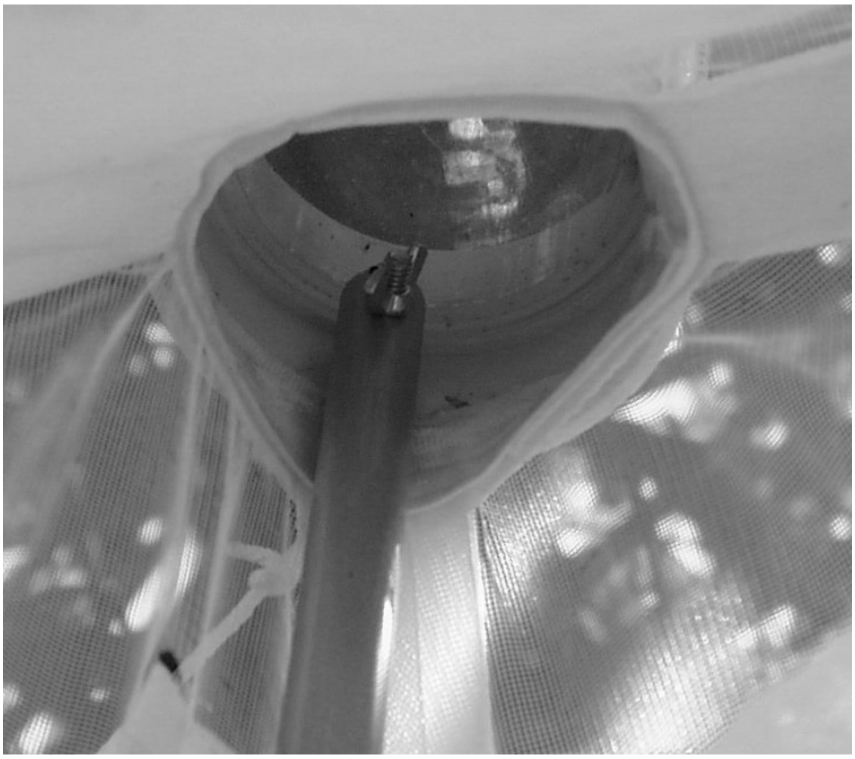


Figure 29. Close-up of connection between collection chamber and Malaise trap.

A Malaise trap captures insects that fly onto the vertical panel of mesh. Insects that land on the panel will walk to the top of the net and become trapped in the roof eave-like upper area where the top of the trap meets the vertical panel. Insects aren't always the smartest organisms. The efficiency of a trap can be enhanced by devising a collection chamber that attaches near the top of the net (Figures 27 & 29).

Using the photographs as a guide someone with good sewing skills can construct a trap very similar to the one illustrated in which one end of the "roof" is higher than the other end and fitted with a collection chamber. A collection chamber can be devised from plastic pipe with a funnel inverted inside to trap organisms in the chamber. However, a very simple trap can be made with 4 pieces of netting. The dimensions of the completed trap are not critical. Cut 2 identical end pieces similar in shape to the front panel (easily seen in the first photograph) of the trap shown in the photograph. These pieces are shaped like the end of a house with a steeply pitched roof. I suggest that they be at least 5'-6' tall at the peak of the “roof”. The centerpiece is a simple rectangle with the same height as the end panels just described and a length of 6’-8’. Longer lengths are fine but can be difficult to set-up and will be more likely to be blown over or damaged in high winds. I wouldn’t make a trap over 12’ long. Traps shorter than 6’ will not be very effective as a longer trap but are easier to handle. The final piece, the “roof”, is cut to fit the dimensions of your other pieces. Make sure you use seam tape when sewing all of the pieces together. This type of trap will collect a wide variety of flying insects but needs to be checked more often than a trap fitted with a collection chamber because it is easy for insects to escape. A Malaise trap can be used in conjunction with a light trap (described in the next chapter) to collect night flying insects.

Ideas for some simple research projects using a Malaise trap:

1. Construct traps from light and dark material and compare the diversity and abundance of organisms that you collect in each.
2. Place your traps in different habitats and compare the diversity and abundance of organisms you collect. Traps set in forest clearings, old growth forest, secondary growth forest, forest paths, open fields and across streams will all have different species diversity.
3. Yearlong studies at one site can be used to examine seasonal changes in species diversity and abundance. Longer term studies can develop a database that can be used to look for population cycles.

LIGHT TRAPS

Light traps are another simple method of collecting flying insects. The construction of a light trap can be fairly complex with timers and rotating collection chambers or very simple. There are a number of commercially available light traps that do a great job. I have provided photographs (Figure 30) of one type just as a means of illustration. The light trap shown here breaks down for storage in a 5 gallon plastic bucket and has a large aluminum funnel to trap disoriented insects in the bucket. The beauty of this type of light trap is that it can be left unattended and insects will become trapped in the bucket for later collection. You can even place a killing jar or jar of preservative under the funnel to kill insects as they become trapped. The problem with this type of trap is they are expensive to purchase and difficult to fabricate.

The most economical version of light trap is simply a light source. Light traps can utilize any type of light source even a lantern but the nature of the light will influence the "catch". Be very careful using blacklight (UV) sources as you can cause painful and permanent damage to your retina if eye protection is not used. Also remember that some light sources can be very hot. While a simple light trap is very cheap and easy to use it requires the collector to be present and actively engaged in collecting throughout the collection period. On the other hand it allows the collector to focus on certain species or insect groups rather than the broad-spectrum sample collected in a bucket type light trap. The efficacy of the light source can be increased by placing a light source under a tent-like structure formed by draping a white bed sheet over a clothesline and staking out the corners or by placing the light in front of a white sheet hung over a line. It is easier to see and collect insects, especially smaller ones, when they are on a white background. You can also use a Malaise trap in conjunction with a light source. If you use a sheet or Malaise trap in conjunction with your light, remember that some light sources are very hot and could start on fire or melt fabric.

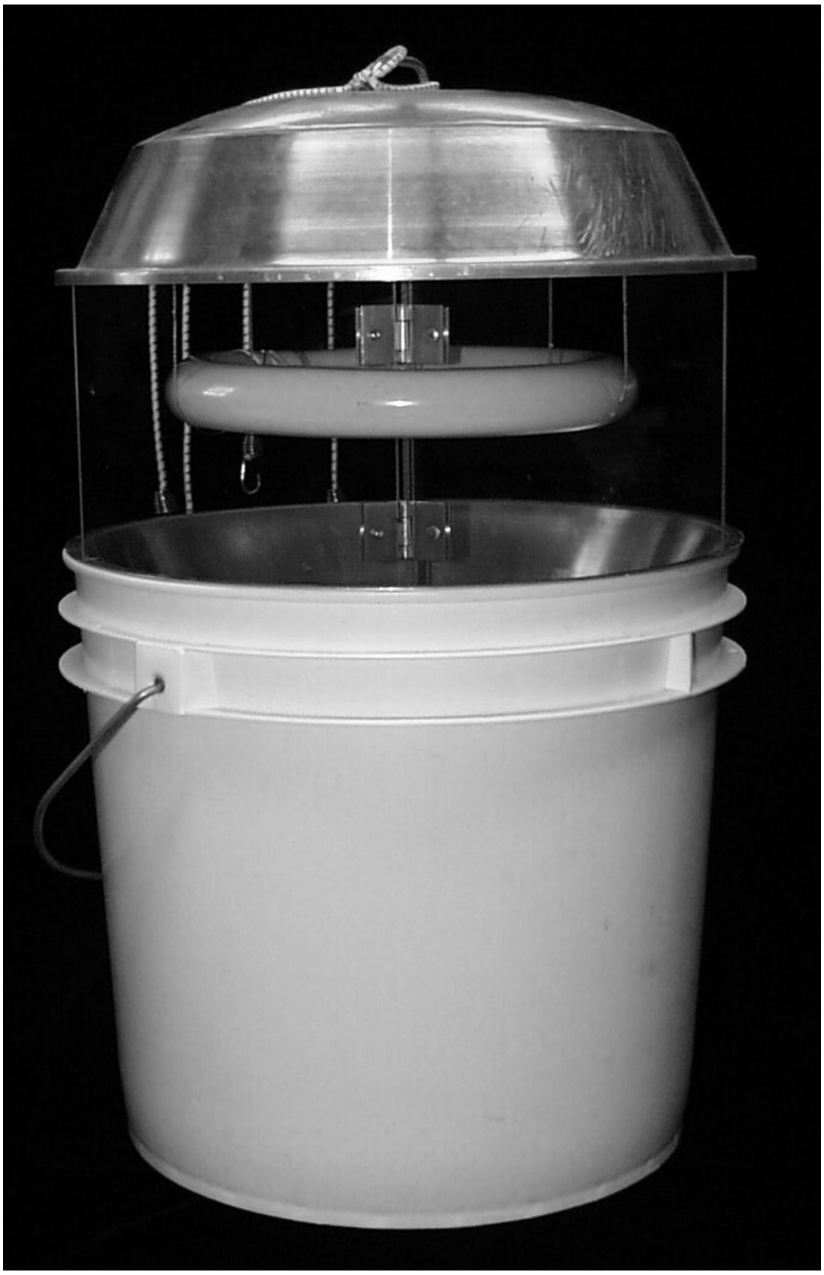


Figure 30. Light trap with bucket collection chamber.

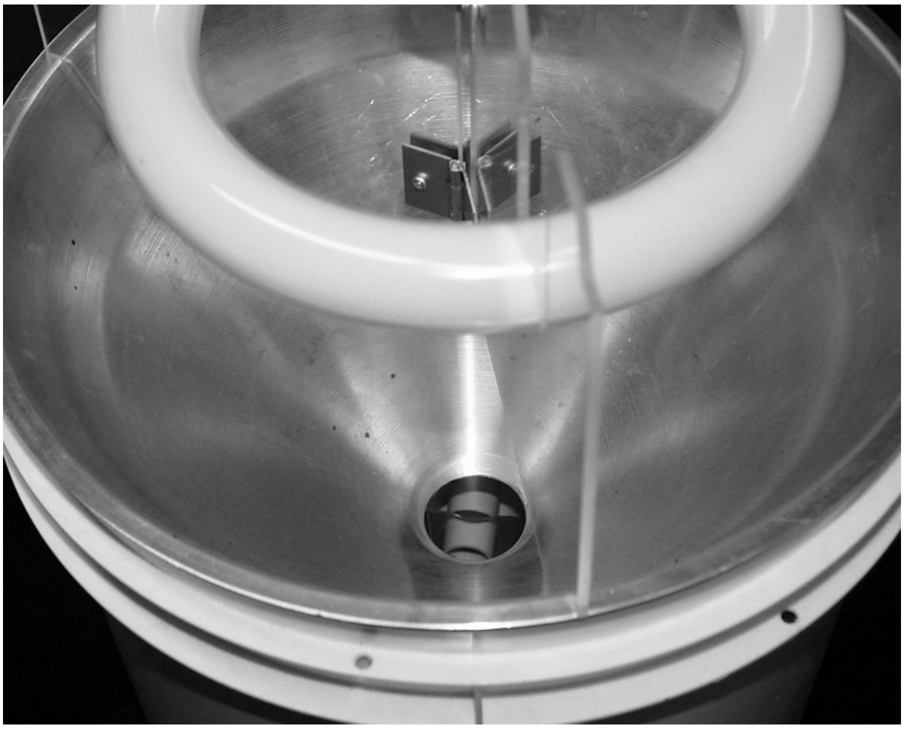


Figure 31. Close-up view of the inside of the light trap shown in Figure 30, funnel directs insects to the inside of the bucket and a collection jar.

Ideas for some simple research projects using a light trap:

1. Place your traps in different habitats and compare the diversity and abundance of organisms you collect. Traps set in forest clearings, old growth forest, secondary growth forest, forest paths, and open fields will have different species diversity.
2. Compare catches using different light sources. You can compare different types of lights such as fluorescent vs. incandescent, lights with different wavelengths (i.e. colors, yellow vs. red, red vs. Uv, colored filters are available that would allow you to be very specific in wavelength), or lights of different intensity.
3. Yearlong studies at one site can be used to examine seasonal changes in species diversity and abundance. Longer studies can develop a database that can be used to look for population cycles.

COVER BOARDS

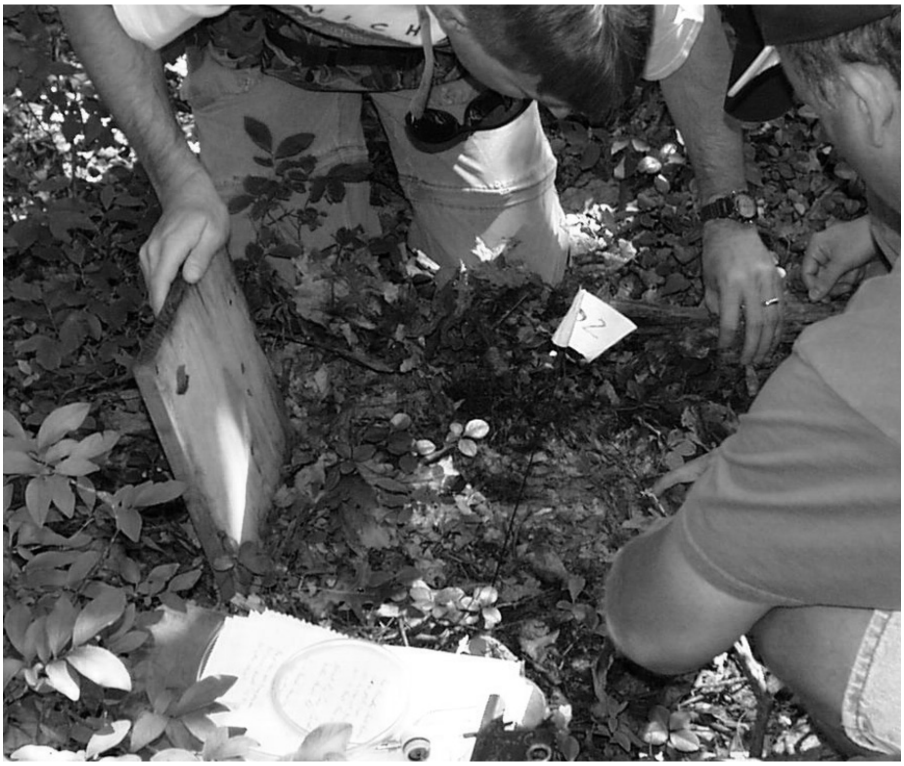


Figure 32. Checking under cover boards for specimens, unlike these students it is safer to lift the board away from you by the far edge in case dangerous organisms are hiding underneath.

Cover boards are an incredibly easy and economical method for carrying out long term studies of ground dwelling organisms. The boards are placed in a habitat and monitored on a set schedule for an extended period of time. There is virtually no set-up time and the cost is negligible. Boards can be constructed from dimensional lumber or masonite but the latter should be painted with polyurethane (or a similar product) in damp environments. Other types of wood products should be avoided because most of them are not very waterproof and many of them will give off formaldehyde that could impact your data by adversely affecting the organisms you are trying to study. Obviously you don't want to use pressure-treated lumber. A cover board should at least 1' square but dimensions are not critical. It is important that all of the boards are of an equal size and composition. If a board is too large it may be difficult to handle and complicate data collection. Data interpretation and reporting is simplified if each board covers a "simple" area, e.g. 1 sq. ft. or 0.25 sq. m, as opposed to 1.23 sq. ft. or 0.18 sq. m. Make sure to mark the location of each board to facilitate relocating them.

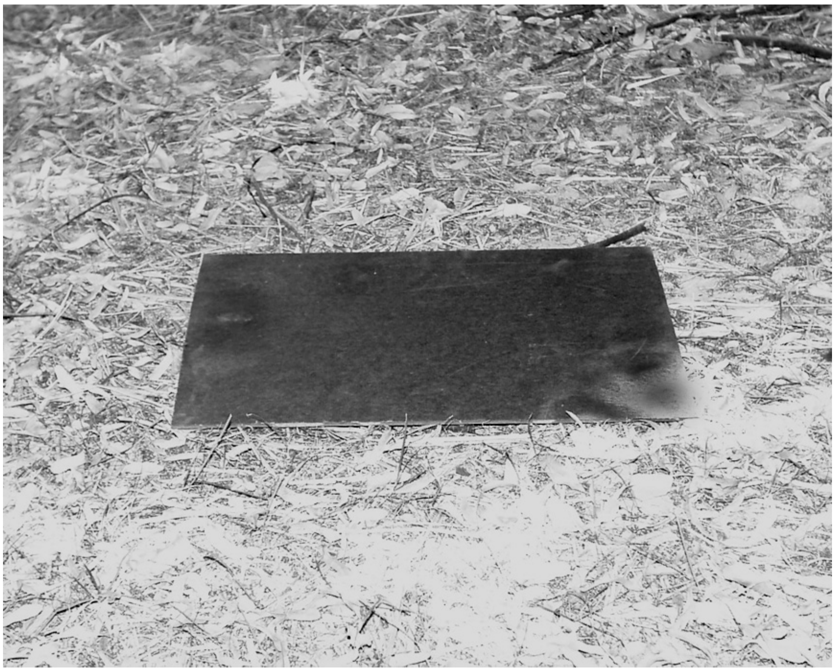


Figure 33. A cover board in place under a tree.

When collecting data it is best to work in teams of 3-4 because organisms will scramble out of sight as soon as a board is lifted. Designate one person as the data recorder and the others as data collectors. For maximum efficiency each collector can specialize in several groups. During each sample period each board is tilted up and the number and kind of organisms under each is recorded. Specimens can be collected for identification in the lab or to retain as voucher specimens. Replace the board in its original location after each observation period. If poisonous snakes or other dangerous organisms are located in the study area, it is a good idea to use a hooked stick to lift a board rather than using your hands. It is also a good idea to tilt the board away from you. If your sampling interval is more than a week, it is a good idea to watch for wasp activity at each board prior to lifting it up to make sure that ground nesting yellow jackets haven't established a nest under a board. During sampling try to keep the area underneath a board in the shade if possible as this will give you more time to count and identify organisms under each board.

Ideas for simple studies using cover boards: In each of these studies try to minimize the variation in the data by making each cover board the same size and using the same number of cover boards for each treatment.

1. Have each student construct several cover boards and place them at home or around the school grounds. Each week, or other time interval, determine what types of organisms are

under each cover board. Ask the students where they think the organisms would be found in nature.

2. Place cover boards in different parts of the habitat (e.g. sunny field, under conifers, under deciduous trees) and compare the types and numbers of organisms you find in each set of cover boards.
3. Place cover boards under different species or varieties of trees and compare the types and numbers of organisms you find under each set of cover boards.
4. Do a long-term study in one habitat. Check the cover boards each week or month. Determine the number and kind of each organism and plot the population density as it changes throughout the year. See if you can predict what will happen in subsequent years.
5. Make several sets of cover boards of different colors. Place them in the same habitat and determine if the color of the board influenced what was found underneath (dark colors will absorb more heat).
6. Make several sets of cover boards of different sizes. Place them in the same habitat and determine if the size of the cover board influenced what was found underneath.
7. Compare the density and diversity of organisms found under a cover board to that found under natural materials such as flatrocks or tree bark. Make sure to compensate for differences in the area covered.

STICKY TRAPS

Sticky traps are commonly used to monitor populations of flying insects especially agricultural pests. When coupled with chemical attractants/messengers, called pheromones, sticky traps can be fairly species specific and are used to help determine when control measures should be used against damaging pests. Sticky traps come in many shapes and sizes ranging from a card to a roofed structure. While sticky traps are economical to produce and easy to use they do have two major drawbacks, they are a real mess to use and specimens collected on the traps are very difficult to save for later identification or as voucher specimens.

The “stick” of a sticky trap is usually the result of a liberal coating of Stickum’ Special or Tanglefoot. These materials have the appearance of petroleum jelly but are much thicker and very sticky. Both can be purchased at well-equipped garden stores or through several online sources and come in a variety of packaging options. Tanglefoot can even be purchased in an aerosol can but, while very convenient, it is an expensive option. Either product can be applied with a spatula or putty knife or diluted with paint thinner and applied by brush. The latter method allows for thinner, more even coverage of an object. Vaseline can be used as a replacement for these products but will not remain effective for more than a day or so and will trap only very small organisms such as whiteflies and thrips. Wear rubber gloves and old clothing when working with these materials.

Sticky traps are usually yellow or white in color and fall into three broad categories: cards, 3-dimensional objects, and roofed. Card traps can be any size but are generally less than 1 square foot. A variety of materials can be used to make the cards as long as the sticky material you apply remains on the surface of the card rather than soaking into the surface. Heavy coated paper, Plexiglas, and masonite all work well. Avoid corrugated cardboard and poster board, these materials are not very weather resistant and will absorb the sticky material. Durable materials such as masonite can be recycled by scraping the old sticky material from the cards with a putty knife and applying a fresh sticky coating. If the cards are constructed from lightweight material, clothespins and binder clips (bulldog clips) can be used to hold the traps in place when they are distributed in the sampling area. Traps can be clipped directly to twigs and foliage or clips and clothespins can be permanently affixed to a stake or pole. Drill holes in the tops of cards made from heavier material so they can be hung like Christmas ornaments. You can coat one or both sides of card traps with sticky material depending on the nature of the experiment you are performing.

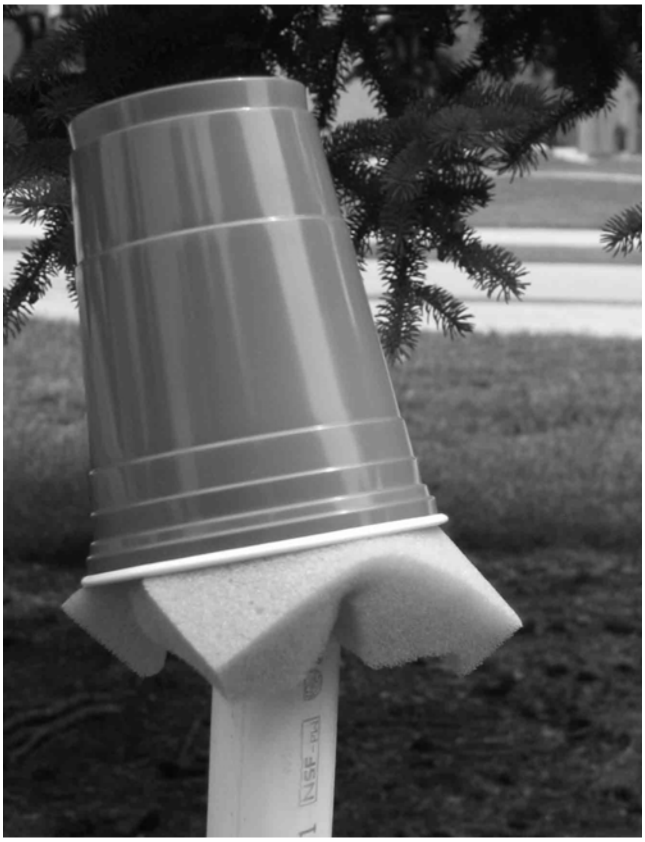


Figure 34. A sticky trap made from a plastic cup. Foam rubber holds the cup in place on a stake made from PVC pipe.

Three-dimensional traps will often attract insects that wouldn't be attracted to other types of traps. For example some types of fruit flies are attracted to round shapes. One inexpensive option is to use 12 oz. plastic cups as traps (Figure 34). Here is an easy way to handle cup-type traps. For each trap you need a 6" piece of 3/4" diam. PVC and a 4" X 4" piece of soft foam (like that used for cushions) approximately 3" thick (measurements are not critical). Cut a slit completely through the center of the foam and insert the PVC into the slit. Invert the cup and push it firmly down over the foam and you now have a cup trap with a handle. It looks like a very odd popsicle or ice cream treat! For easy placement in the field use wooden dowels with a diameter small enough that the PVC will slip over as stakes to hold the traps. If you are using a large number of traps you might want to make a rack out of plywood and wooden dowels to help you move them around. Another type of 3-D trap can be made from styrofoam balls purchased in a craft store. Balls of about 3" diameter are a good size. Use stiff wire such as a large paperclip to hang the balls in the sample area.

Roofed or covered traps (Figure 35) are the most complex of the three types of traps. Several styles of these are available commercially at reasonable prices. Some have replaceable bottoms for prolonged usage. These work best in conjunction with pheromone dispensers. Pheromone dispensers (note rubber septum in close-up of covered trap) are highly species specific and can be purchased for a large number of species. Living females of the species you are interested in collecting can be used in place of a pheromone septum with mixed results. The females must be kept alive so they continue to produce pheromones. They must be gently restrained or confined in the trap area. Covered traps are commonly used in the tree fruit industry to monitor pest insect populations. Due to their complexity it is probably easier to purchase commercially made roof-type traps than it is to construct your own. Cockroach and mouse motels are variations on this type of trap.

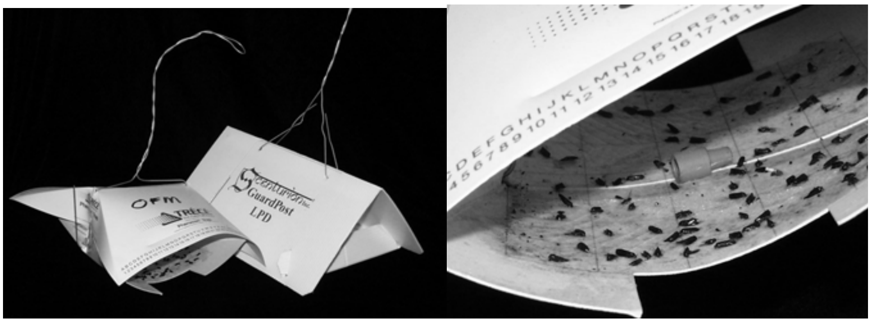


Figure 35. Commercially available sticky traps, note the rubber septum (center right) which disperses pheromones.

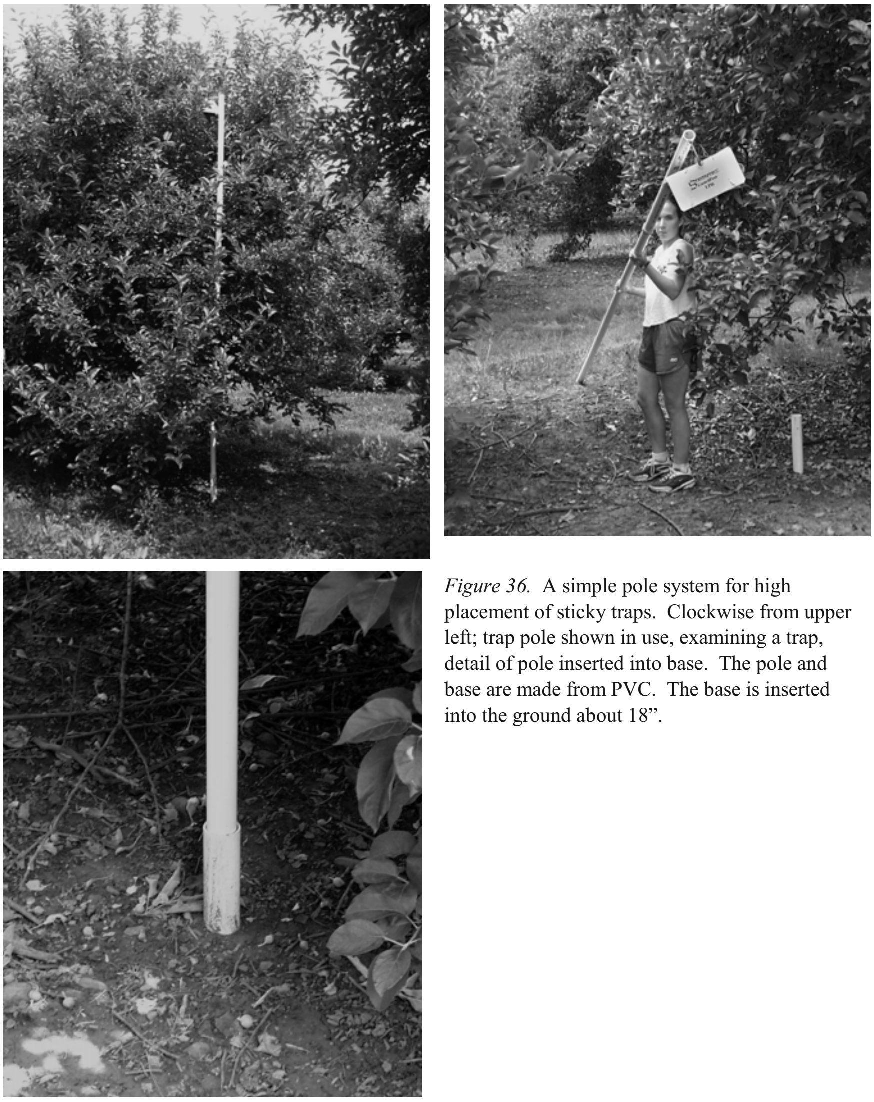


Figure 36. A simple pole system for high placement of sticky traps. Clockwise from upper left; trap pole shown in use, examining a trap, detail of pole inserted into base. The pole and base are made from PVC. The base is inserted into the ground about 18”.

Ideas for simple studies using sticky traps: Unless it is part of your experimental design try to minimize the variation in the data by making each trap the same size and using the same number of sticky traps for each treatment. You need to check the traps on a regular basis, weekly is a good interval. Rather than trying to count and identify organisms in the field it may be easier to bring the traps to a laboratory for examination. Exchanging used traps with new each collection period facilitates this operation and assures that there are always traps in the sample area.

1. Have each student construct several sticky traps and place them at home or around the school grounds. Each week, or other time interval, collect the traps and determine the number and types of organisms on each trap.
2. Place sticky traps in different parts of the habitat (e.g. sunny field, under conifers, under deciduous trees) and compare the types and numbers of organisms you find in each set of traps.
3. Place sticky traps under different species or varieties of trees and compare the types and numbers of organisms you find in each set of traps.
4. Do a long-term study in one habitat. Check the sticky traps each week or month. Determine the number and kind of each organism and plot the population density as it changes throughout the year. See if you can predict what will happen in subsequent years.
5. Make several sets of sticky traps of different colors. Place them in the same habitat and determine if the color of the trap influenced what was collected. This works especially well with ball type traps, paint some normal fruit colors (e.g. red, orange, green) and others odd colors (e.g. blue, black).
6. Make several sets of sticky traps of different sizes. Place them in the same habitat and determine if the size of the trap influenced what was collected. Again ball traps may yield interesting results. Make sure all the traps are the same color. Use ping-pong, tennis and softball sized balls. You can also try larger sizes to see if a “supernormal stimulus” elicits greater responses than “normal” stimulus.
7. Place traps at different heights (e.g. 6”, 1’, 3’ and 6’ above ground) to determine if trap height influenced what was collected. Photographs at the end of this unit illustrate a pole system that allows you to place traps fairly high without the need for a ladder to examine them.
8. Divide cup traps into four quadrants of equal size place them in the study area so they are all aligned. Determine if direction influenced trap catch.
9. Place traps in the foliage of one type of tree. Divide each tree into North, South, East, and West quadrants. Hang traps in each directional quadrant and determine if direction influenced trap catch. Similarly each tree could be divided into low and high canopy or inner and outer canopy to determine if there are differences in trap counts in each area.

AQUATIC TRAPS – LEAF PACKS

Aquatic traps are an interesting method of monitoring and collecting invertebrates in aquatic ecosystems. The fins of the trap provide shelter and refuge for aquatic organisms. When a trap is examined after being left in place for several weeks there will be a diversity of organisms found in the trap. These traps are available commercially but they are also easy and economical to produce at home. To construct a trap similar to the one shown in the photograph you need a 1/2" diameter eyebolt (the one in the photograph is 6" long but the length is not critical), lots of washers, 2 nuts that fit the eyebolt, a sheet of 1/8" masonite, and marine grade urethane (or similar product).

Prior to cutting the masonite give it several good coats of marine grade urethane, a paint roller will facilitate completion of this step. Alternatively you could use a waterproof material, such as plastic, instead of the masonite. I prefer masonite because it is inexpensive and easy to cut and drill. Cut the masonite into 6" square pieces, these will form the "fins" of the trap. You will need 12-15 fins/trap. Drill a hole in the center of the masonite pieces sized to accommodate the eyebolt. Touch up the ends and holes of the masonite with urethane to assure that your trap is waterproof. Thread a nut all the way to the eye end of the bolt.

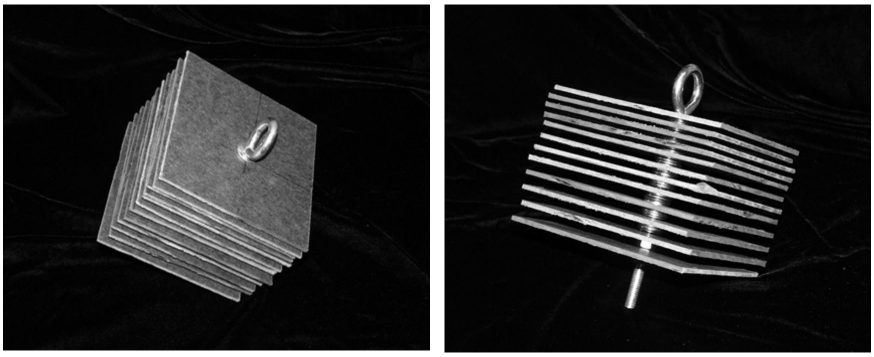


Figure 37. Aquatic trap using waterproofed masonite squares, the space between the pieces will influence what is caught in the traps.

Follow with a washer. Place your first fin after the washer and then alternate washers and fins until your trap is the desired size. The distance between the fins will influence what is caught in the trap so you may need to experiment to make sure the trap fin spacing is appropriate for your study. You should probably put at least 2 washers between each fin. The trap in the photograph has the fins set up in two different spacings (look closely at the right hand photograph) for a simple experiment designed to examine the influence of fin spacing on number and diversity of organisms collected.

To use your traps simply place them in an aquatic environment for a predetermined time period. Usually 2-4 weeks will produce good results. Secure the trap with a length of strong nylon cord attached to the eyebolt and anchored on shore. When it is time to evaluate the contents of the traps take them apart over a white pan. Use forceps, a large pipette or turkey baster, and a paintbrush to collect organisms from the pan.

Leaf packs are a variation of this trap and will produce very similar data although a leaf pack will attract organisms interested in eating the leaves as well as colonizers. To make a leaf pack you will need a good supply of soaked leaves. You can use freshly collected green leaves or older leaves collected from the bottom of a pond or stream. You will probably get different results with each type of leaf. The species of tree will also influence your results. You will also need lightweight fishing line and a heavy sewing needle. Take 15-20 leaves (the number is not critical but use the same number in each leaf pack), lay them flat on top of each other and loosely sew them together with the fishing line. Just a few stitches are all you will need for each pack. Remember that for a leaf pack to work small organisms need to be able to squeeze between the leaves so do not sew the leaves too tightly.

While these two types of traps yield similar data there are pros and cons of each. The aquatic trap allows you to easily determine the exact area of trap surface available for colonizers. Your data can then be expressed in units such as number of caddisflies/sq. mm. This cannot be easily calculated with a leaf pack. Leaf packs are cheaper and easier to produce but are not reusable. Leaf packs work best in slow or non-moving water, they are difficult to secure in place in moving water. Aquatic traps will work in almost any type of water condition. It is impossible to precisely control the spaces between the leaves of a leaf pack this could have a small influence on what is collected. The distance between fins in aquatic traps can be precisely adjusted.

Ideas for simple studies using leaf packs and aquatic traps in each of these studies: try to minimize the variation in the data by making each trap the same size and using the same number of traps for each treatment.

1. Place traps in different parts of the habitat (e.g. riffle, run, pool) and compare the types and numbers of organisms you find in each set of traps.
2. Do a long-term study in one habitat. Replace the traps each week. Determine the number and kind of each organism and plot the population density as it changes throughout the year. See if you can predict what will happen in subsequent years.
3. Determine the influence of fin spacing on the number and diversity of organisms collected in one habitat. Do this by building traps with more than one fin spacing, similar to the one shown in the photograph. In this way you can be more certain that any differences you find are due to the spacing of the fins and not the variance in location of traps i.e. if you have two traps each with different spacing they will not be in exactly the same location in the habitat.
4. Compare the number and diversity of organisms collected in leaf packs produced from fresh versus aged leaves or from different species of trees.

BURLESE FUNNELS

Burlese funnels are used to separate organisms that live in soil, litter, mast and other detritus from the material in which they live. They are a great way to collect small cryptic organisms that would be difficult to collect using other methods. Unlike many other collection techniques to use a Burlese funnel you sample a unit of habitat rather than targeting organisms. Burlese funnels come in many shapes and sizes. Larger funnels allow more material to be placed in the sample chamber and processed at one time. Material placed in a Burlese funnel slowly dries out forcing organisms living in the sample to move deeper down into the material. Eventually the organisms fall out of the bottom of the sample, into the funnel where they are directed into a jar of preservative. Using a low wattage light bulb to shorten the drying time of the sample can facilitate the process.

A small Burlese funnel can be made from a two liter soda bottle (Figure 38) and a small piece of hardware cloth (1/8" or 1/4" mesh works best). Cut the bottle into two pieces just below the centerline. Cut a piece of hardware cloth that fits in the upper section of the bottle approximately at the point where the bottle starts to curve. Invert the top section of the bottle into the base of the bottle. Pour a few ounces of preservative (50-70% alcohol) into the base. Carefully place your sample on the hardware cloth. You may want to do this before you place upper portion of the bottle in base as small bits of sample material may fall through the cloth and contaminate the preservative making it harder to work with the sample. You can use a gooseneck lamp to help dry out the sample. It will take about a week for the sample to be processed. You may want to cover the top of the apparatus with a piece of fine mesh screen to prevent organisms from exiting the top of the funnel. Use a dissecting microscope to aid in the examination of the preservative because many of the organisms you collect will be $\leq$ 1mm long.

A soda bottle Burlese funnel is economical, easy to make, and easy to use. However there may be times that it is advantageous or necessary to process large amounts of material. In those circumstances larger Burlese funnels are required.

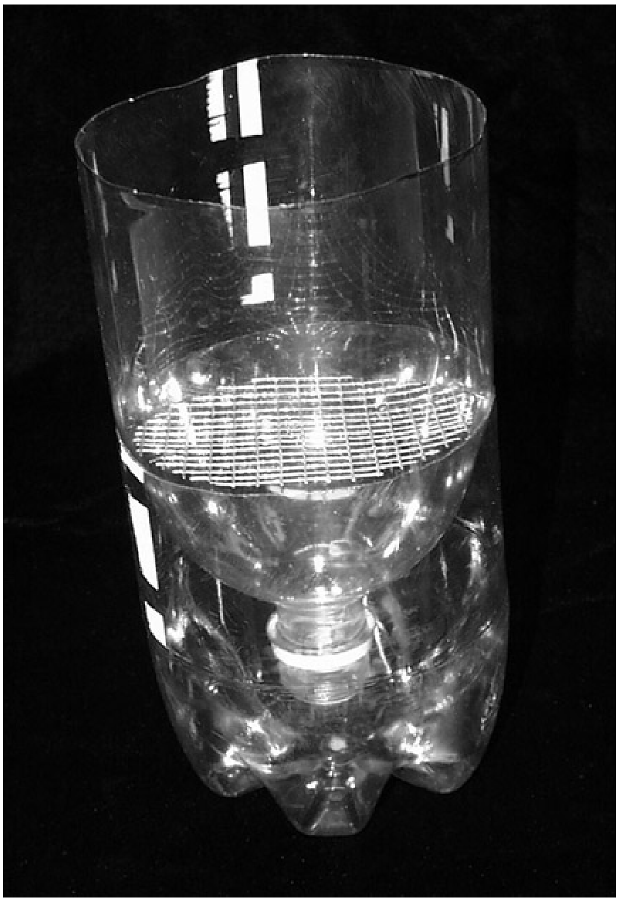


Figure 38. A Burlese funnel made from a soda bottle. Alcohol should be placed in the base to kill insects that fall from the sample.

To construct a mid-size Burlese funnel (Figure 39) you will need two large coffee cans (small cans may also be used), hardware cloth (1/4" or 1/8" mesh), aluminum flashing or a funnel with a diameter slightly larger than the coffee can, an aluminum clamp light, and 4 short pieces of 1" X 2" lumber. Cut both ends out of one of the coffee cans this is the sample holding chamber. Leave one end intact in the other can it forms the base of the apparatus to hold the funnel and the preservative. Use lumber to construct a small square wooden frame with an inside dimension just slightly larger than the diameter of the coffee can. The frame should fit snuggly over the large opening of your funnel. Cut a piece of hardware cloth to fit the frame and staple it in place. If you do not have a funnel that fits your needs construct one out of aluminum flashing. Hold the flashing together with several short nuts and bolts (pan head). Duct tape will also work. Seal the inner seam of the funnel with duct tape. Your funnel should be about 3/4 the height of your coffee can.

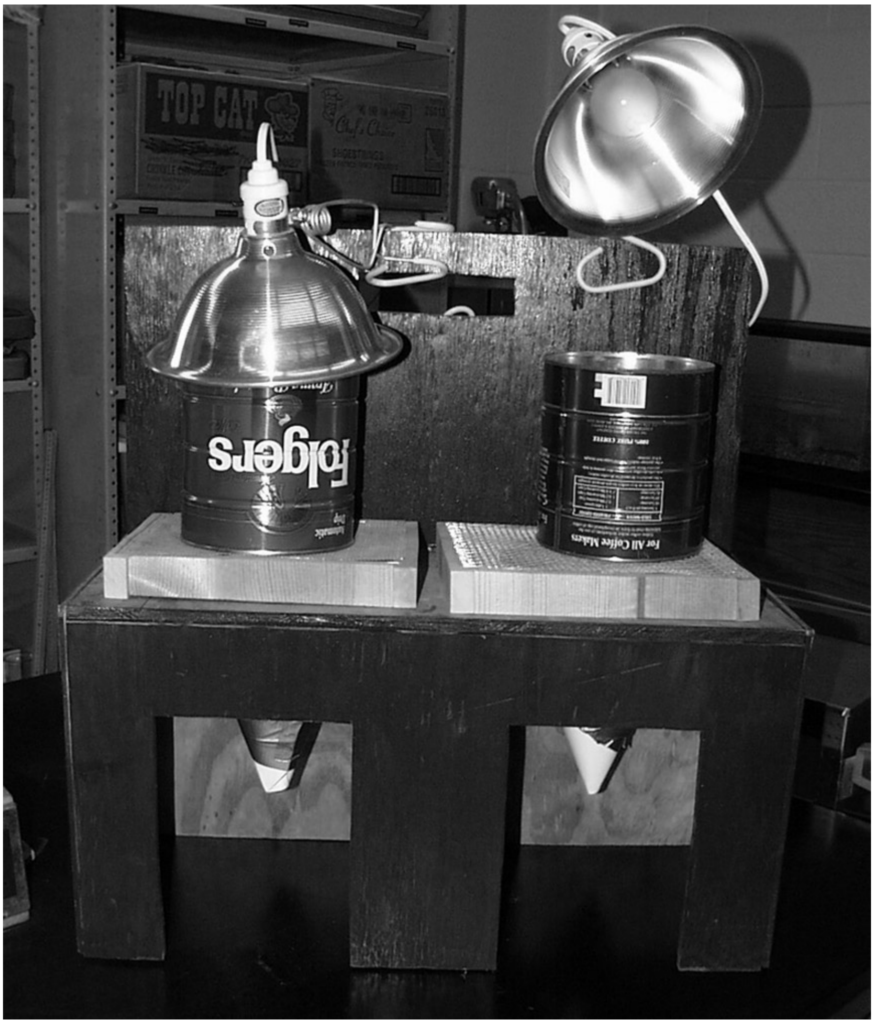
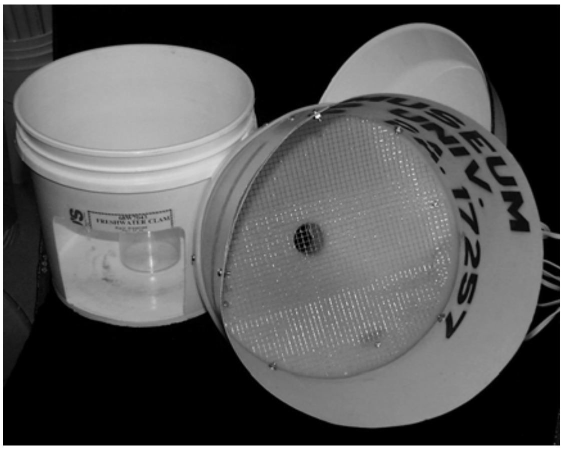


Figure 39. Burlese funnels made from coffee cans. Collection jars containing alcohol should be placed under the funnels.'

To assemble the apparatus place the can with the closed end on your work surface. Put a small amount (about 1/4") of preservative directly into the can or place a small baby food-type jar containing preservative into the can. Place your funnel on top of the can; the rim of the can supports the funnel. If you are using a container other than the coffee can to hold preservative make sure the end of your funnel is directed into the container. Place the wooden frame over the funnel and place the open-ended can on top of the frame. The clamp light rests on top of the can. Make sure you have a low wattage (20 watts is good) bulb in the light. Your Burlese funnel is ready to go! If you intend to use large numbers of this type of funnel construct stands out of plywood to more securely hold the Burlese funnel, provide easier access to the preservative chamber and provide a location for lights to be clamped.

Even a large coffee can may not provide a large enough sample chamber to meet your needs. A large Burlese funnel (Figure 40) can easily be made from a large plastic funnel, aluminum flashing, hardware cloth, a 5-gallon plastic bucket, and a light fixture. The bucket acts as a stand for the funnel and houses the container of preservative. Cut a large opening in the lower side of the bucket to allow easy access to the collection jar. The funnel needs to support the weight of a large sample so it is better to purchase a durable funnel with a diameter of 12”-15” rather than fashioning one out of flashing. Place the funnel square and straight on top of the bucket and draw a line around the funnel at the point where it meets the bucket rim. Just above and along this line drill three holes around the circumference of the funnel that are equidistant from each other. Put a short nut (it needs to project from the funnel side about 1/4”) and bolt through each hole. These are designed to hold the funnel steady and prevent it from slipping around on top of the bucket.

Fashion a sample chamber out of flashing. The lower edge of the chamber fits inside the top of the funnel. The chamber should be about 12” high. Cut a piece of flashing to the height you desire and two times the circumference of the funnel. Fashion a tube out the flashing that is two layers of metal thick. Use several short nuts and bolts to hold the flashing tube together. Attach the chamber sides to the funnel with several nuts and bolts. Cut a piece of hardware cloth that fits snugly against the sides of the chamber. With a Burlese funnel this large it is imperative to use a light to help dry out the sample material. The light fixture shown in the photograph was salvaged from a construction site but a suitable light can be located at a large hardware store. A light socket mounted on a wooden bracket will also work.



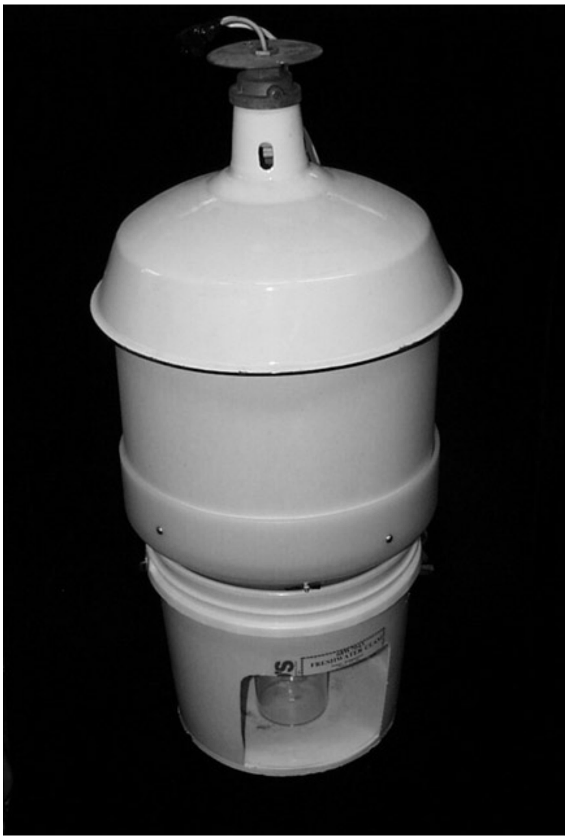


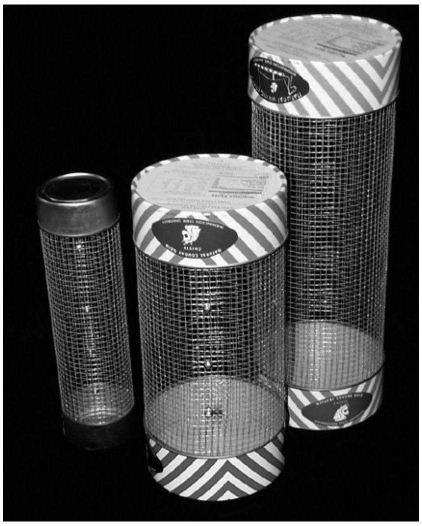
Figure 40. A large Burlese funnel made from a plastic bucket, aluminum flashing and an old light fixture.

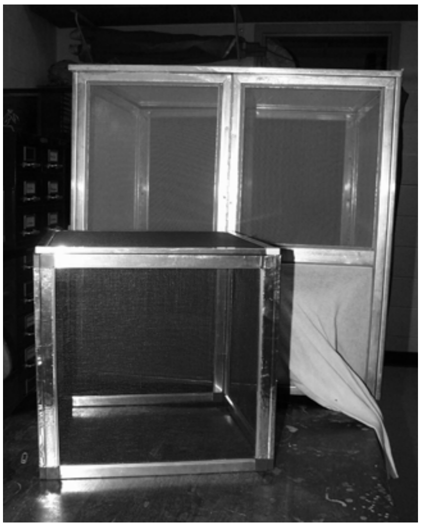
Ideas for simple studies using a Burlese funnel: in each of these studies try to minimize the variation in the data by collecting samples of the same size and collecting the same number of samples for each treatment.

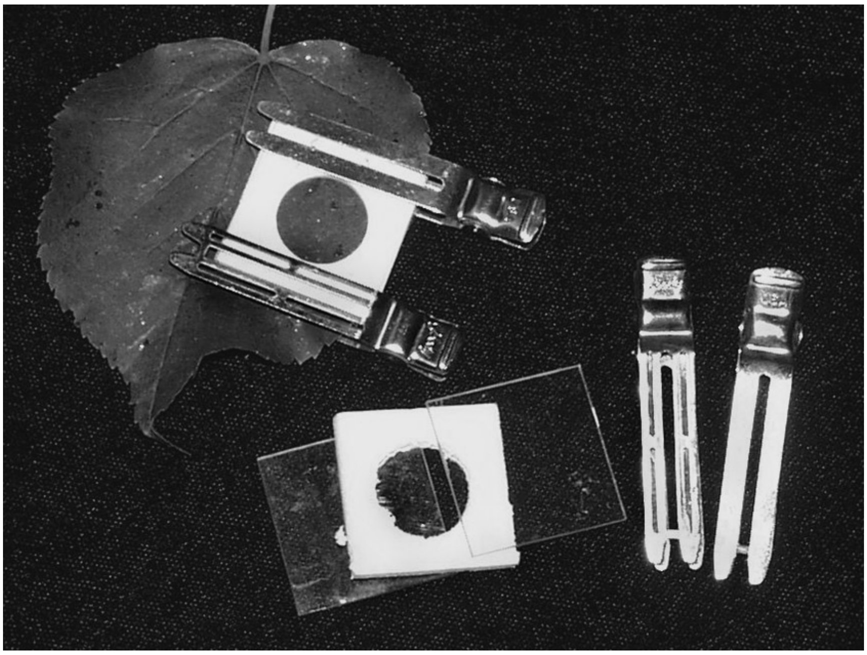
1. Collect samples from different niches in an ecosystem e.g. grassy area, leaf litter, sunny vs. shaded area.
2. Do a long-term study in one habitat. Determine the number and kind of each organism and plot the population density as it changes throughout the year. See if you can predict what will happen in subsequent years.
3. Have each student make a soda bottle funnel.
4. Many kinds of material can be processed in a Burlese funnel: soil, leaf litter, galls, pinecones, flowers, etc.

CHAPTER 4

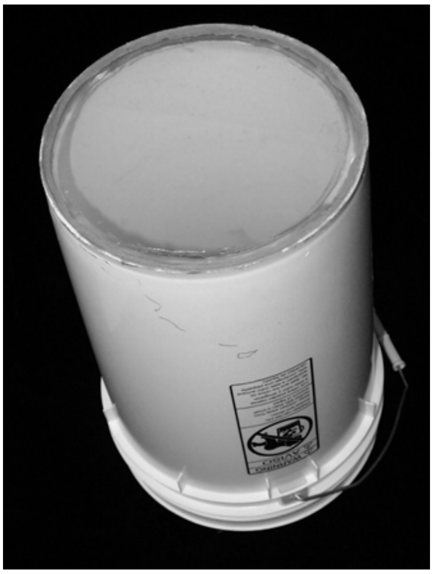
CAGES AND OBSERVATION ARENAS







BUCKET VIEWERS



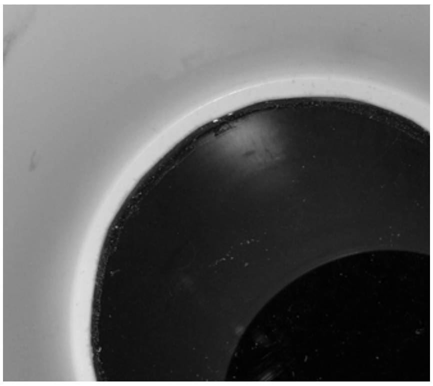


Figure 41. Glass bottomed bucket, inner construction detail (above) shows the lip upon which the clear pane rests.

In the field it is sometimes difficult to see what is transpiring below the surface of the water and it is not always prudent or feasible to use a diving mask or goggles. In some instances a glass bottomed bucket viewer can provide a window into the underwater world.

To construct a viewer you will need a 5-gallon bucket and a piece of glass or plexiglas (recommended). Cut the bottom out of the bucket, leave a lip about 1/2” wide around the circumference (see photograph above). The lip will provide extra support for the clear pane. Cut a piece of plastic or glass sized to fit inside the protruding rim on the bottom of the bucket and rest flat on the lip. Note that the clear pane is mounted on the outside of the bucket bottom so pressure exerted against the pane when the viewer is used will not cause the pane to come loose and leak. Use a strong adhesive and silicone caulk to affix the clear pane in place and seal it in place. Use caulking on both sides of the pane. As soon as everything is dry the viewer is ready to be used. Push the bottom of the bucket a few inches under the surface of the water and look through the top. You will be able to see underwater just as if you were wearing a diving mask. You can protect the clear pane from scratches and breakage by cutting the lower 5”-6” from the bottom of another bucket and using it like a lens cap for your bucket. The bottom can also be used to hold samples and examine specimens that you collect using your viewer.

SLEEVE CAGES

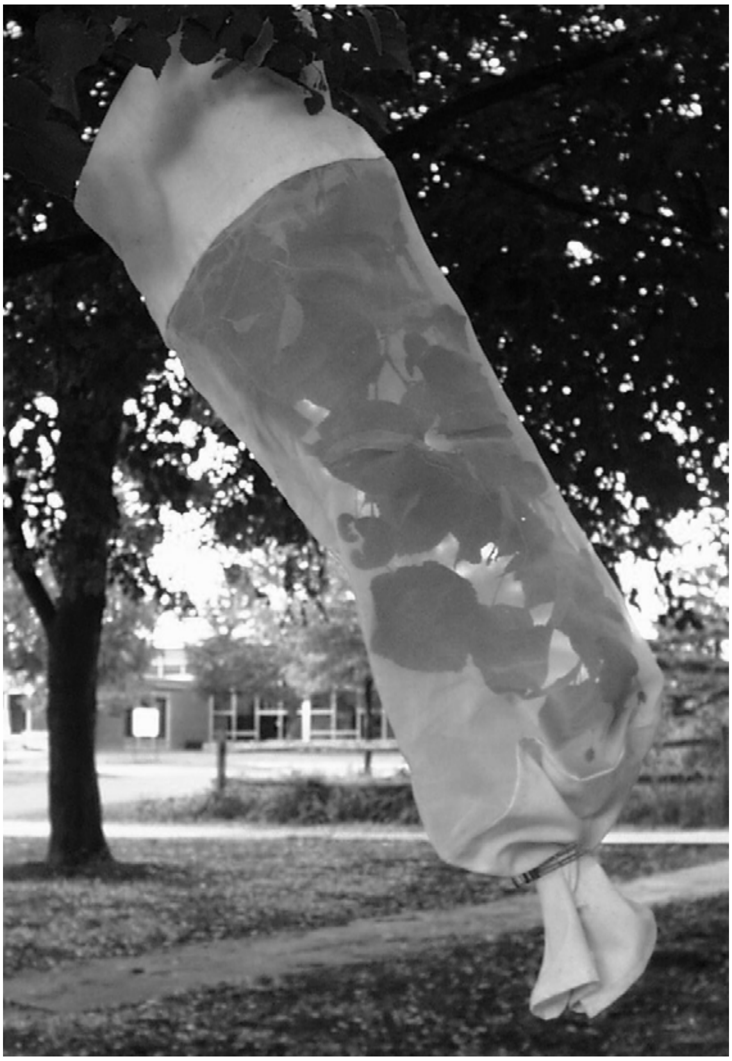


Figure 42. Sleeve cage enclosing the end of a tree limb.

Sleeve cages are used for rearing organisms on a branch or twig. When organisms are inside of a sleeve cage they are effectively confined to one area of a plant and prevented from coming into contact with other organisms. Sleeve cages can be made from many types of material but weather resistant fabrics are the best choices. The mesh size of the sleeve can be critical depending on the reason the sleeve cage is being employed. Tighter weaves form the most effective barriers to movement in or out of the sleeve but they also impact the microclimate of the habitat by restricting airflow and holding in water vapor. Carefully consider the pros and cons of the material you choose.

As the name would imply a sleeve cage is a piece of mesh fabric sewn into a tubular, sleeve-like form. Nylon organdy is a good choice of fabric for the mesh portion of the cage but heavier screening material, even house screen, can also be used. Adding several inches of heavy canvas or nylon to each end of the sleeve provides a durable area for tying off the ends of the sleeve when it is in use. The dimensions of the sleeve can be adjusted to meet your needs. In some instances it may be important minimize contact between enclosed vegetation and the sleeve. In those circumstances (e.g. some Homoptera produce copious amounts of honeydew which can "gum up" the sleeve) X-shaped cross braces made from thin wooden dowels or tongue depressors can be attached to the branch to hold the sleeve above the leaves.

Applications for a sleeve cage:

1. Use sleeve cages as rearing cages for herbivores. If you are rearing a large herbivore, such as a silk moth caterpillar, the sleeve cage and herbivore may need to be relocated to new branches as leaves are devoured.
2. Enclose colonies of aphids, whiteflies and other Homoptera inside a sleeve cage to trap emerging parasites.
3. Do a study on the effect of predators or parasites on herbivores (aphids are a good choice) by using sleeve cages to exclude natural enemies from one treatment of your experiment. A simple experiment would have 3 treatments: closed sleeve cages, open sleeve cages and no sleeve cages. Be sure to replicate the treatments, 10 of each are a good number. Leave the cages in place for a set time interval, at least several weeks, after which the sleeves are removed and populations in each treatment are examined. Analyze your data with a paired t-test or a simple one-way ANOVA.

CLIP CAGES

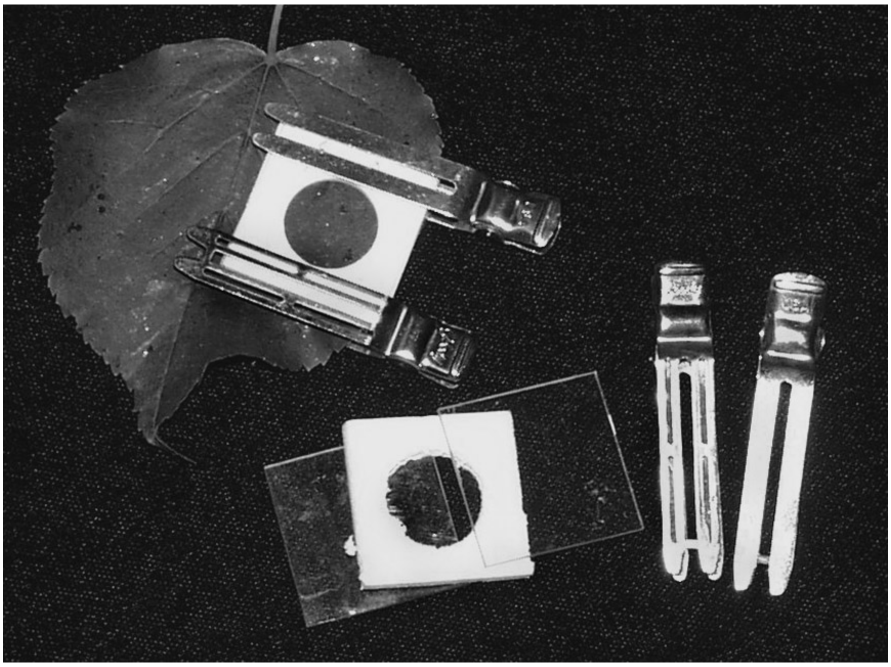


Figure 43. Clip cage components and assembled cage on a leaf (upper left).

Clip cages are a great way to confine small organisms for behavioral studies. For each clip cage you need two pieces of 1” X 1” X 1/8” thick plexiglas, two metal hair clips (don’t use binder clips as they will damage the leaf), and one piece of 1” X 1” X 1/8” thick high density foam (Dr. Scholl’s foot pads work well). Use a cork borer to cut a hole through the center of each piece of foam to form the observation arena. Slightly bend the top prongs of each hair clip so the prongs rest flat against the top piece of plexiglas when the cage is in place. To use the cage place the hole in the foam around the area of interest on the leaf surface. Place one piece of plexiglas on the opposite side of the leaf directly under the foam and place the other piece of plexiglas on top of the foam. Use two clips to hold all of the parts in place. The leaves can remain in place on the plant or they can be removed and taken to a laboratory.

Applications for clip cages:

1. Use the clip cage to form an observation arena to study feeding, parasitism, predation, mating, etc. in small organisms. Clip cages easily fit under a dissecting microscope to facilitate observation.
2. Enclose eggs, larvae, or pupae in clip cages to determine what emerges or hatches from each. Small parasitic wasps can often be collected from eggs and immature stages of small insects such as aphids and whiteflies.

ALUMINUM SCREEN CAGES

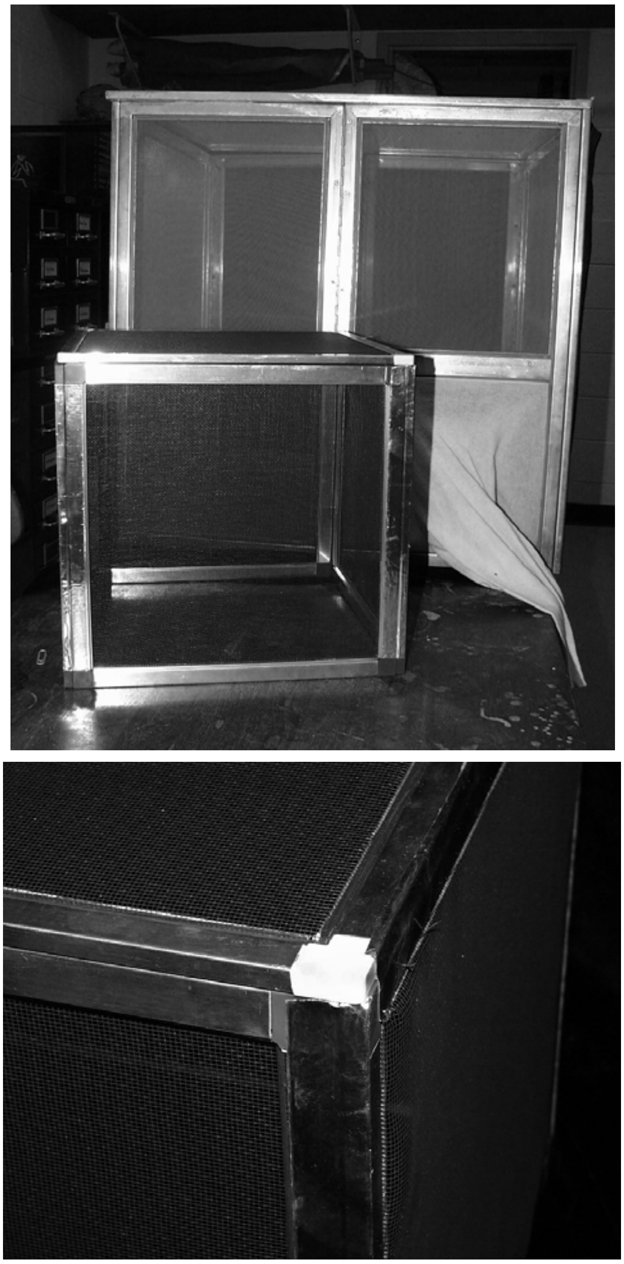


Figure 44. Cages constructed from aluminum screen frames (top) the sides of the larger cage are made from several subunits, corner detail (bottom).

Small cages such as the sleeve and clip cages described previously can be very useful but at times larger cages are needed for rearing and holding organisms in the laboratory. There are several types of commercially produced cages that are durable and effective but they are also expensive. Cages of almost any size can be made from household aluminum screen framing material (also used to build beating trays, see page 19). Screen frame is sold in most hardware stores in lengths up to 8’ in various colors and sizes. The cage on the previous page is made from plain aluminum 5/16” frame. Plastic corners and spline (be sure to buy the correct size) are sold separately. The frame can be easily cut with a hacksaw to the desired length.

To build a cage you need enough framing material, including corners and splines, to make 5 screened sides, mesh fabric (as always, remember to carefully consider mesh size when making your choice), sheet aluminum for the bottom (masonite and plexiglas will also work), a roll of aluminum tape, and a few 1/2” sheet metal screws. I recommend that the top panel be sized to rest on top of and flush with the side panels (see detail, Figure 44). Once each side is assembled place a piece of mesh material over the frame and, starting in one corner, push the spline into place with the handle of a screwdriver. Use one piece of spline for the entire frame, cut it after you have finished pushing it into place. If the material (e.g. metal screen) is heavy you may have difficulty pushing the spline into place. After the spline is entirely in place trim the mesh material close to the frame with a razor blade. You can assemble the entire cage using sheet metal screws. (remember to drill pilot holes in the metal) but as an alternative use aluminum tape, this material is strong and seals each corner better than sheet metal screws. Tape only one side of the top panel using the aluminum tape to form a hinge. Use sheet metal screws on the other three sides.

If you are building a large cage (>3’/side) it is better to use additional framing pieces to add support. T-shaped plastic connectors can be used to add crosspieces. Alternately construct each side of the cage from several sub-units (Figure 44). One useful variation of the basic design described here is the addition of an access sleeve (see large cage in Figure 44). This allows access to the inside of a cage without opening it completely up, a handy attribute if you are rearing noxious or nasty insects. A sleeve can be easily constructed out of tightly knit somewhat elastic fabric such as T-shirt material. A sleeve from an old shirt works well. You will probably need to subdivide the side of the cage that has the sleeve such as in the photograph at the start of this unit. The sleeve material is held in place with spline.

One last alternative construction is to assemble a cage without a bottom. The cage can be placed over a plant in a greenhouse or field for rearing organisms on a host plant. If a cage of this design is used outside it is best to stake it down so it is not blown over in high winds.

BUCKET REARING CAGES

Durable and versatile cages can be constructed from 5-gallon buckets. I'll explain several variations. Bucket lids can be fitted with a rigid viewing window made from Plexiglas, glass, or screening. Leave the heavy rim and seal area of the lid intact. Cut the middle portion out of the lid leaving a 1/2" lip in the flat portion of the lid just inside of the rim. Save the middle portion of the lid for later use. Screening can be glued directly to the lip using a hot glue gun. Remember to carefully consider mesh size before gluing the screen in place. If you prefer a clear pane instead of screen, cut a piece of glass or plexiglas so it will rest on the lip. Use glue to hold the pane in place and then carefully seal the pane, inside and out, using silicone caulking. Use either type of lid with a bucket as a very simple cage. This is not a good type of cage for highly mobile or flying insects because the lid must be completely removed to water and feed the insects. A sleeve can be easily added to increase the usefulness of a bucket cage.

To make a bucket cage sleeve you will need the middle portion of the bucket lid or a similar piece of flexible plastic, a sleeve from an old shirt (stretchy jersey material is best), and 6-10 nut, bolt and washer sets (approximately 1/2"long). You can also sew a sleeve from fabric if you don't have an old shirt to sacrifice. Cut an 8" – 9" diameter hole in the side of the bucket about 6" from the bottom of the bucket. Cut the old bucket lid center into a ring with an outer dimension about 1" diameter greater than the diameter of the bucket hole and an inner dimension equal to the diameter of the bucket hole. This ring will act like a giant washer to help hold the sleeve in place. Line up the ring with the hole in the side of the bucket and drill a series of evenly spaced holes through the ring and the bucket around the perimeter of the hole sized to fit the bolts you plan on using. Drill the first hole at the top of the ring and secure it in place with a nut and bolt. Then drill the bottom hole and secure the ring to the bucket with another nut and bolt. After you have the top and bottom of the ring secured to the bucket it is much easier to assure that the remaining holes you drill will line up during assembly. After all of the holes have been drilled remove the ring from the bucket and fold the sleeve over the ring. Again starting at the top, attach the ring and sleeve to the bucket. Use washers to help prevent tearing the sleeve. Make small holes in the sleeve with an awl or punch to facilitate pushing the bolts through the fabric.

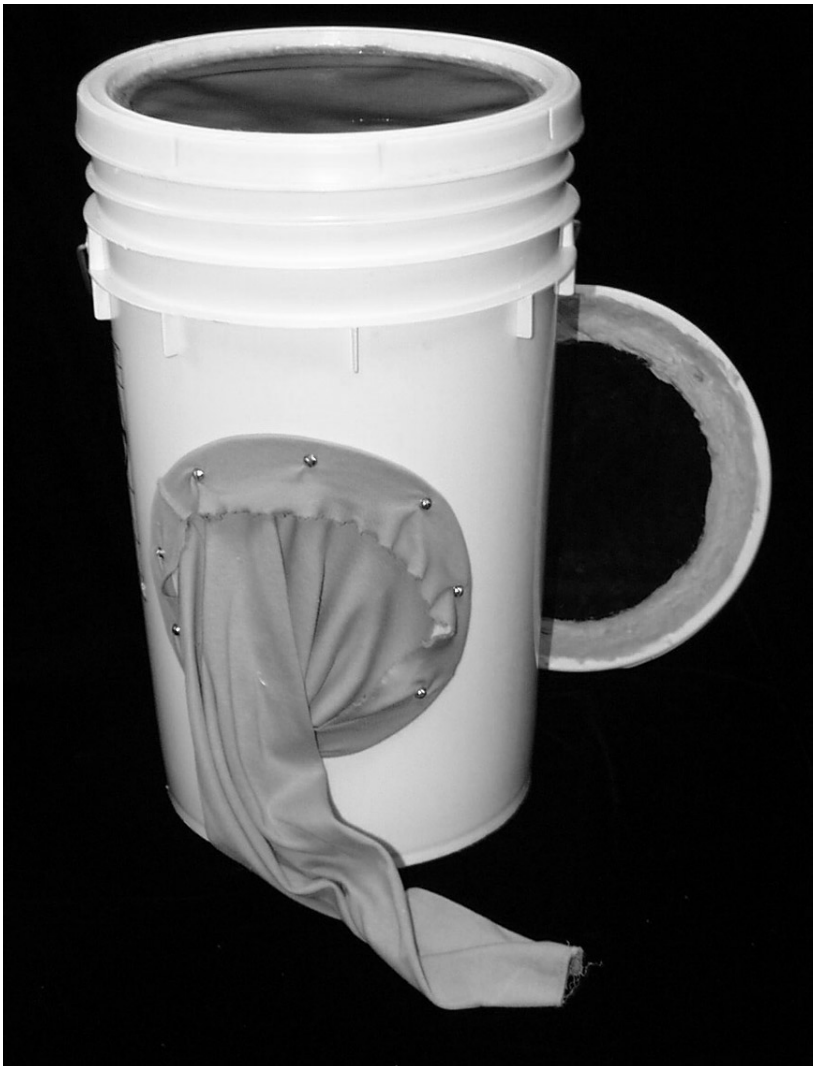


Figure 45. Bucket cage with screen top (optional clear pane top shown in background) and sleeve access.

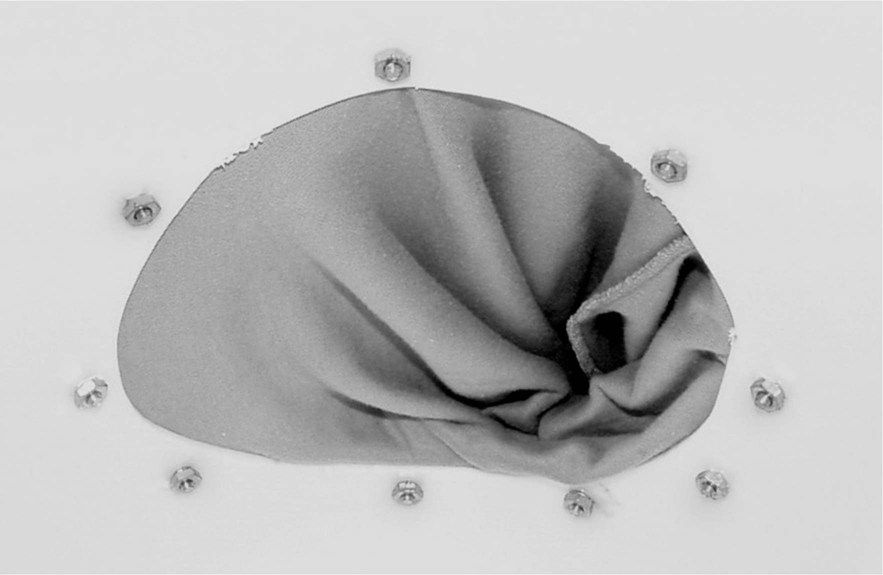


Figure 46. Detail of the inside of a bucket cage showing attachment of sleeve to the side of the bucket.

The sleeve will minimize escapes from the cage while allowing full access to the inside. As you retract your hand from the sleeve flap the fabric slowly and softly to prevent insects from following your hand. Tie a loose knot in the sleeve as close to the bucket as possib when it is not being used.

Another variation of the bucket cage is designed for outdoor use especially with plants. With the bottom removed a bucket can be placed over a small plant and pushed several inches down into the soil to seal the lower edge. Once the bucket is secured a screened or clear paned lid completes the cage. This type of cage is good for studying herbivorous insects on their host plants. You can also use the cage to enclose objects allowing you t collect insects from decomposing material such as walnuts, acorns, feces, carrion, etc. an other sources. You can also you a sleeve on this type of trap.

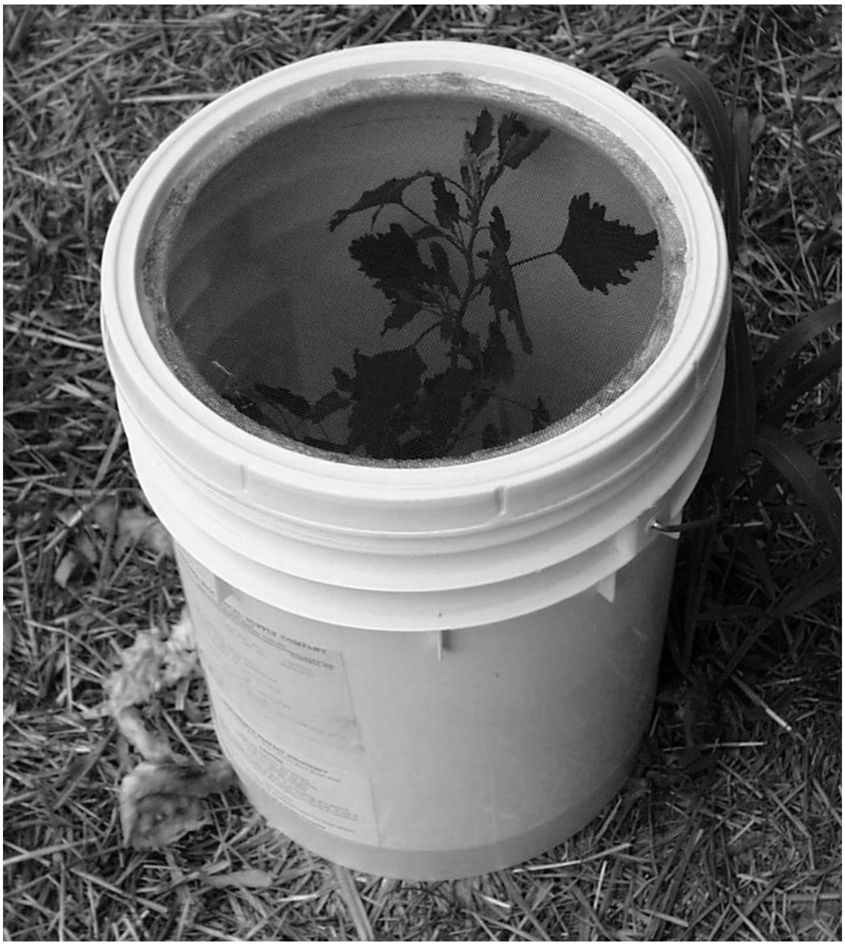


Figure 47. Bucket cage enclosing a plant, the bottom edge of the bucket is pushed tightly into the soil.

PVC PIPE CAGES

PVC pipe can be used to build a frame or scaffold to support netting for a cage. Very large cages can be made using this material. The cages are also easy to disassemble, transport and store when not in use. To build a cage from PVC pipe you will need to purchase corner couplers and, depending on the size of your cage, T and + shaped couplers as well. For cages less than 3 feet in all dimensions 3/4" PVC pipe can be used but 1" pipe is more durable. The difference in price between the two different sizes of pipe is negligible. To build larger cages you should use 1" pipe and you should add extra horizontal and vertical support members approximately every three feet. Usually the coupler/pipe fittings are fairly tight and the components will stay together on their own. If the frame is intended for long-term use, glue the frame components together with standard PVC glue but if you would like to be able to disassemble the frame use nuts and bolts to connect the components.

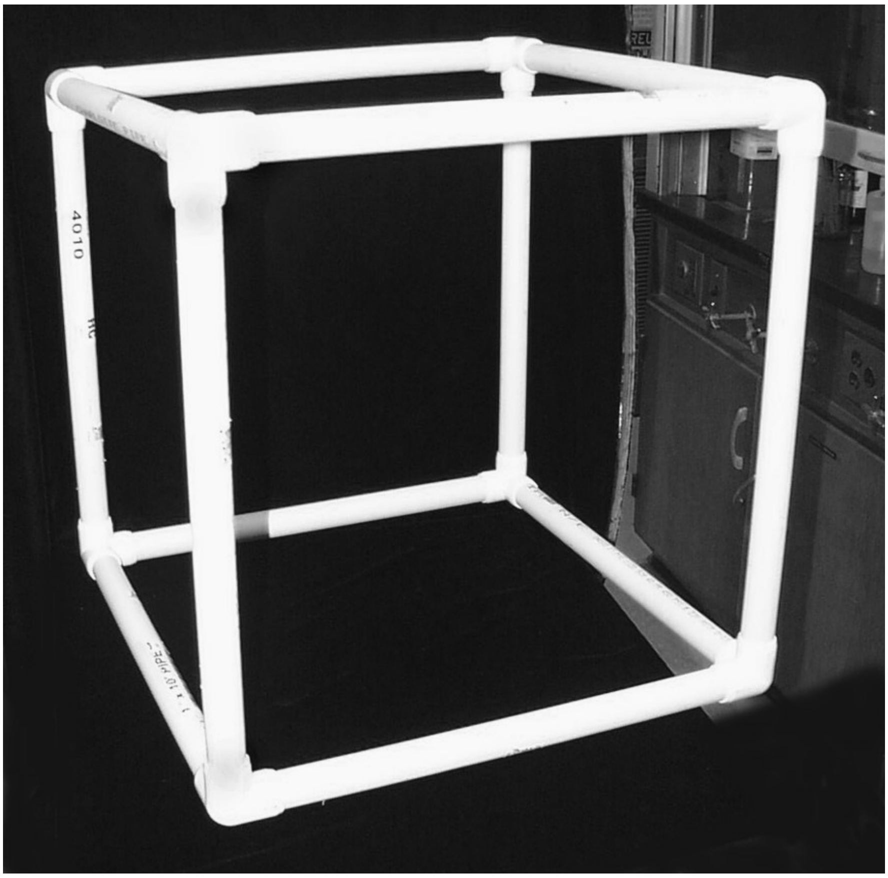


Figure 48. Frame for PVC cage. Very large cages can be made using this type of construction.

Screen for the cages can be made from a variety of materials such as nylon organdy, household screen or greenhouse shade cloth. For small cages the screen can be sewn as a large bag with a drawstring into which the frame is inserted. Tie off the drawstring to seal the cage. This approach works well for cages up to about a 3 foot cube. For larger cages it is best to imagine the cage is an odd tent. Drape the screen over the frame and stake out the bottom margin to form a seal. Cages can be constructed that will cover an entire small tree or a row of plants in a field.

CAN AND WIRE CAGES

Cages for holding larger insects can be made out of tin cans and hardware cloth. For each cage you will need two identical cans, a piece of hardware cloth (1/4" mesh is best) and several sets of nuts, bolts and washers. Form a cylinder from hardware cloth with a diameter just slightly smaller than the inside diameter of the cans you are using. Leave about 1" overlap at the seam. The cloth may be sharp and pokey so you might want to wear gloves when doing this project. Use short bolts with nuts to hold the wire cylinder together. Use washers on both sides of the wire to prevent bolts from being pulled through the mesh. The cans slip over the ends of the completed cylinder. This type of can is great for holding large insects such as praying mantids, hornworms, and silkworms. To make a really large cage of this type use the bottom portions of 5-gallon buckets instead of cans.

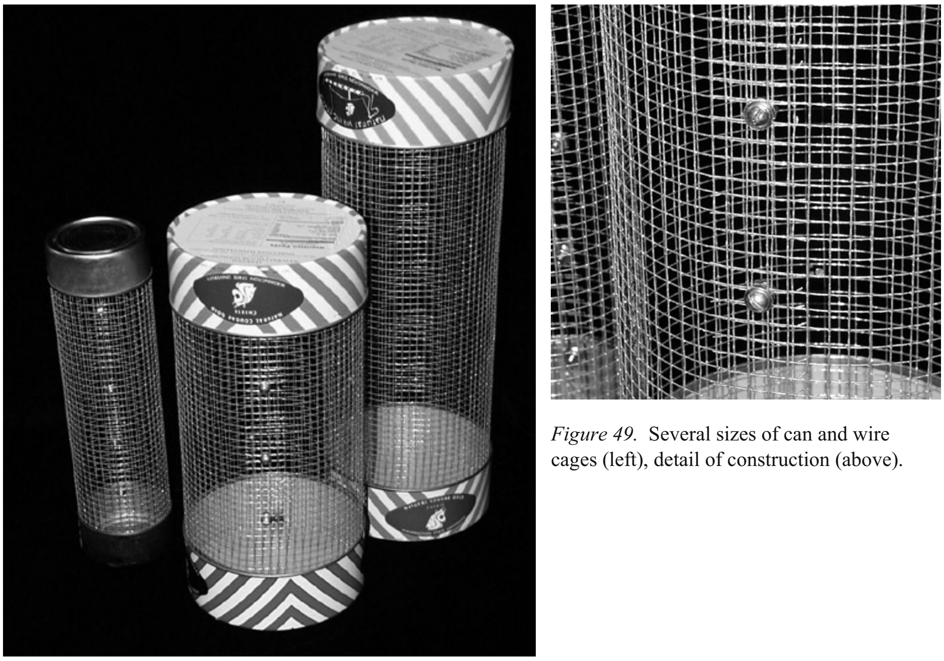


Figure 49. Several sizes of can and wire cages (left), detail of construction (above).

A VARIETY OF SMALL CAGES

There is often the need for small cages when working with insects. In this chapter I will explain how to use a variety of materials to make simple cages. Needlepoint frames can be fitted with nylon organdy, cotton gauze or any of many other materials to be used as covers for a variety of containers. Smaller frames fit nicely over the mouth of large jars and large frames fit over specimen or goldfish bowls. Film canisters (35mm) can easily be modified to function as cages. Use a utility knife to cut the center portion out of the snap cap. You can also cut a window in the side. Use a hot glue gun to affix screen or mesh material over the opening. Most photo processing stores are willing to give you all the canisters you need for free.

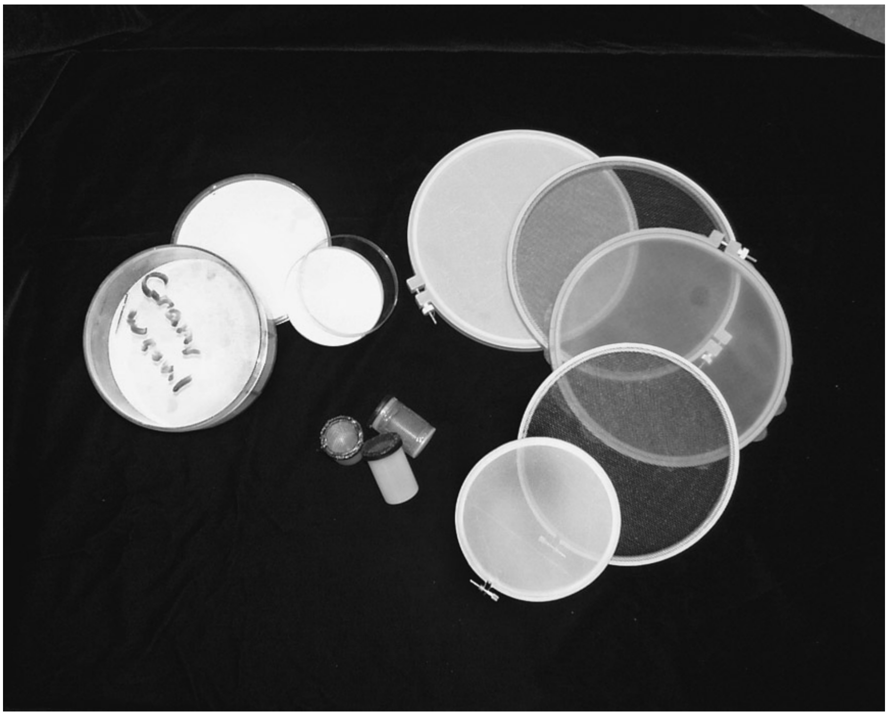


Figure 50. A variety of small cages, clockwise from right, needlepoint frames, film canisters, petri dishes.

Petri dishes come in several sizes and styles. They can be purchased in bulk (usually by the gross) for a reasonable price. They do not seal very tightly so small organisms can escape if you aren't careful. To provide ventilation, holes can be made easily using a heated cork borer. Use a hot glue gun to affix screen over the holes. Pour a thin layer of plaster of paris or casting plaster into the bottom to absorb excess moisture or to help keep the humidity inside the container high. Activated charcoal (available at aquarium stores) can be added to the plaster to absorb CO_2.

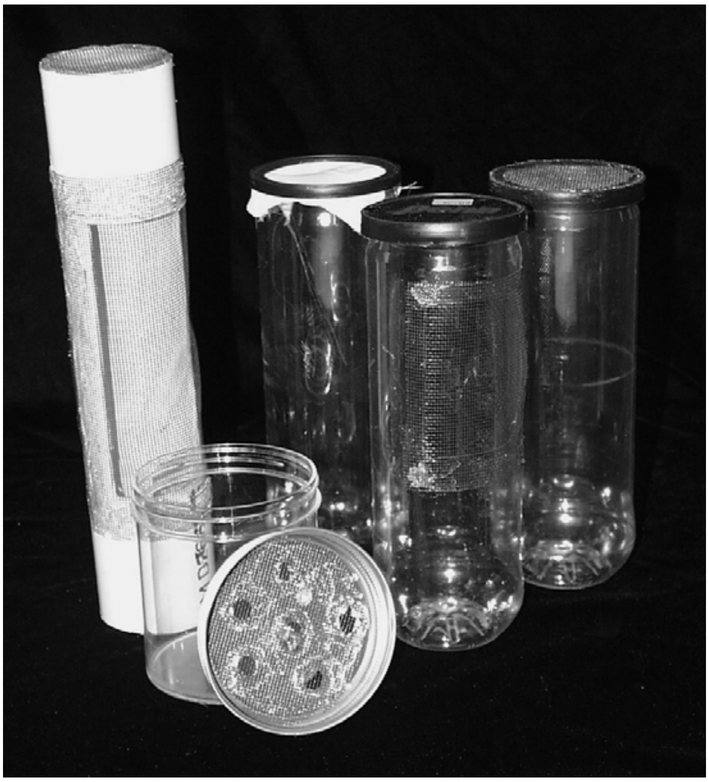


Figure 51. Cages made from (clockwise from right) tennis ball containers, a small jar, and a length of PVC pipe.

PVC pipe can be used to make a cage to enclose a plant. 1” diameter pipe was used to construct the cage in Figure 51 but any size can be used. Cut an opening in the side of the pipe and affix screening over it and the top of the pipe with a hot glue gun. Slide the completed cage over a seedling or shoot. Push the bottom edge of the pipe down into the soil to form a seal. Use this type of cage to either confine insects on a plant or to prevent insects from contacting a plant.

Tennis ball (racquetball containers will also work) containers can also easily be modified into cages. They are especially good containers for aquatic organisms and can act as mini-ponds. As with the film canisters a screen can be affixed to the side or top. Two liter soda bottles can also be used in this way by cutting the top from the bottle. Needlepoint frames make good covers for soda bottle cages. Almost any type of jar can be used as a cage. Cut holes in the lids with a hot cork borer or a drill and affix screening on the inside of the lid (see the container in the lower center of Figure 51). Ball-type canning jars are useful because they have a wide mouth and the center portion of the lid is easily replaced with screening.

SMALL AQUATIC VIEWING ARENAS

A handy little arena for photographing and viewing aquatic organisms can be constructed from microscope slides and lantern glass ("regular" glass can also be used). The arena shown in Figure 52 is about 4" square. The sides are made from lantern glass (available from scientific supply catalogs) but other types of glass also work. Lantern glass is thinner than normal glass, 1/16" versus 1/8". If the arena is to be used for videography or photography, it is important to minimize its depth so it is easier to keep the subjects within the depth of field of the lens being used. The sides and bottom of the arena are made from standard microscope slides cut in half lengthwise. For smaller organisms cut the microscope slides in quarters lengthwise. Use silicone caulking to hold the components together. Plexiglas can be used in place of glass but it is susceptible to scratching which ruins its usefulness for photographic applications.

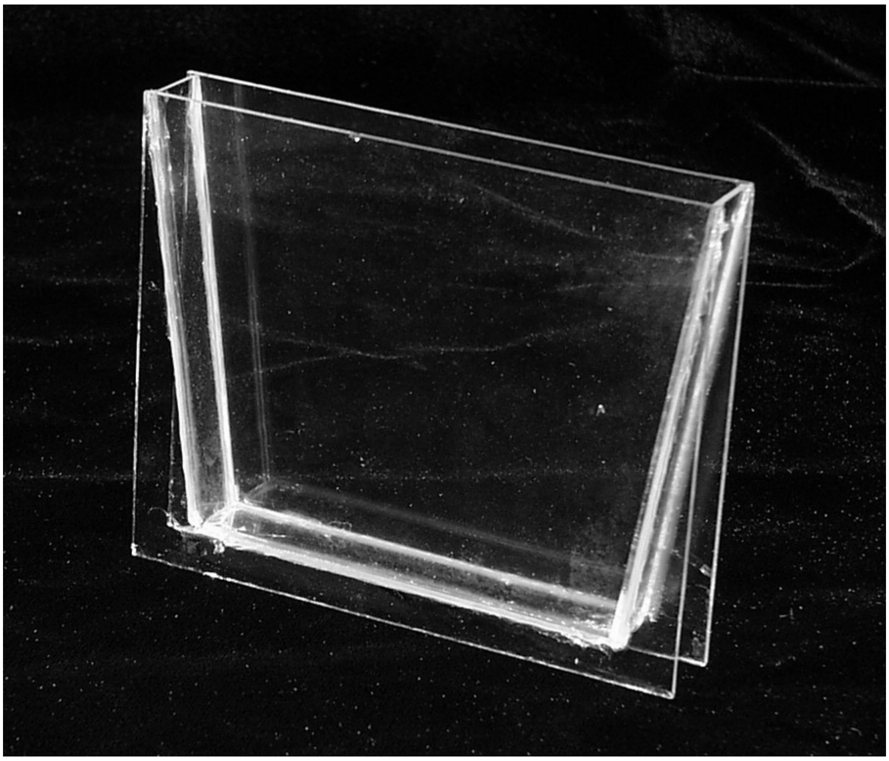


Figure 52. Mini aquarium constructed from lantern glass and microscope slides.

LARGE AQUATIC VIEWING ARENAS

A large aquatic viewing arena can be constructed from plexiglas (1/4” thick) and tygon or vinyl tubing (1/2” outside diameter). You will also need a large number of nuts, bolts and washers (number of sets = width in inches + 2(height in inches)/1.5). The arena shown in Figure 53 is approximately 15” high by 24” wide but larger arenas are easily constructed. For arenas larger than that in the photograph 1/4” thick plexiglas must be used but 1/8” thick plastic can be substituted for smaller projects.

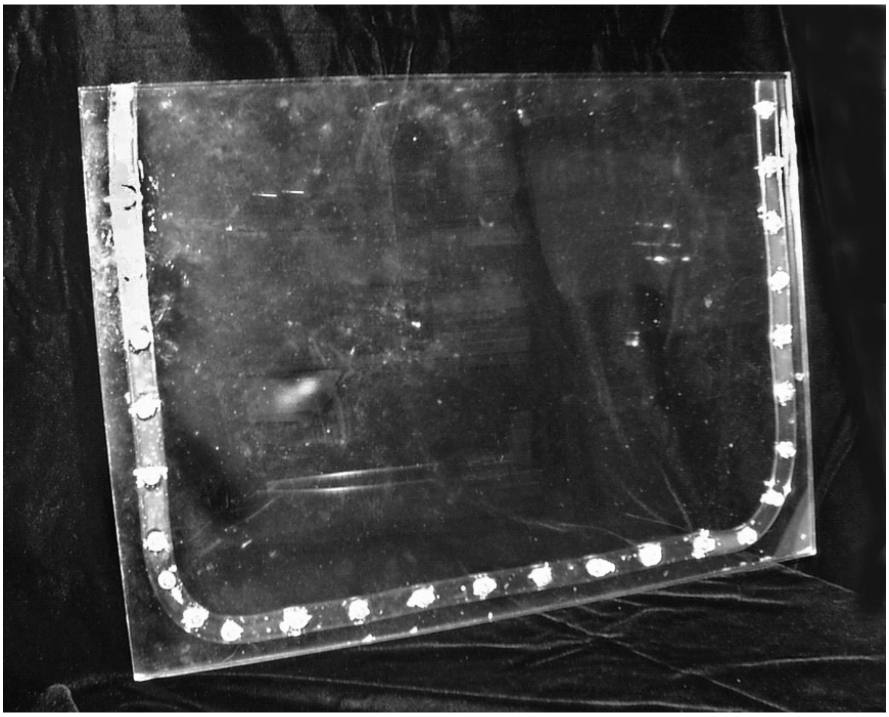


Figure 53. A large aquatic viewing arena constructed from plexiglas. Vinyl tubing forms a seal around the perimeter.

Line up the edges of two pieces of plexiglas previously cut to the size of the desired arena. Place a piece of tubing between the plexiglas sheets as shown in Figure 53. Starting at the top of one side approximately 1 1/2” from the edges of the plexiglas drill a hole, sized to fit your bolts (1/8” or 3/16” diameter are good sizes, 1 1/2” long) through both sheets of plexiglas and the tubing. Insert a bolt with a washer through the first hole and secure it with another washer and nut to stabilize the pieces. At this point the nut should only be snug on the bolt not completely tightened. Make sure the tubing is straight and drill another hole through the plexiglas and tubing near the bottom of the arena side just above where it curves at the corner. Secure this hole with hardware. Continue around the perimeter of the arena drilling only enough holes at this time to secure the plexiglas and tubing into its final configuration (see the photograph). Remember to keep holes at lest 1 1/2” from any edge of the plexiglas.

Once the initial set-up is completed go back around the arena drilling a hole every 1 1/2”. After all of the hardware is inserted tighten the nuts, a socket wrench and ratchet will speed this process. Starting at one end of the tubing tighten each nut slightly moving in sequence from one end of the tubing to the other. Repeat the process, tightening the nuts only slightly during each progression. Tighten the nuts until the arena can hold water without leaking. The inside dimension of the completed arena will be about 1/4”. If a thicker arena is desired use 2 pieces of tubing or tubing with greater diameter and/or thicker walls. Cut off the protruding end of the bolts with a hacksaw being careful not to leave sharp edges.

GLASS-SIDED TERRESTRIAL OBSERVATION CAGES

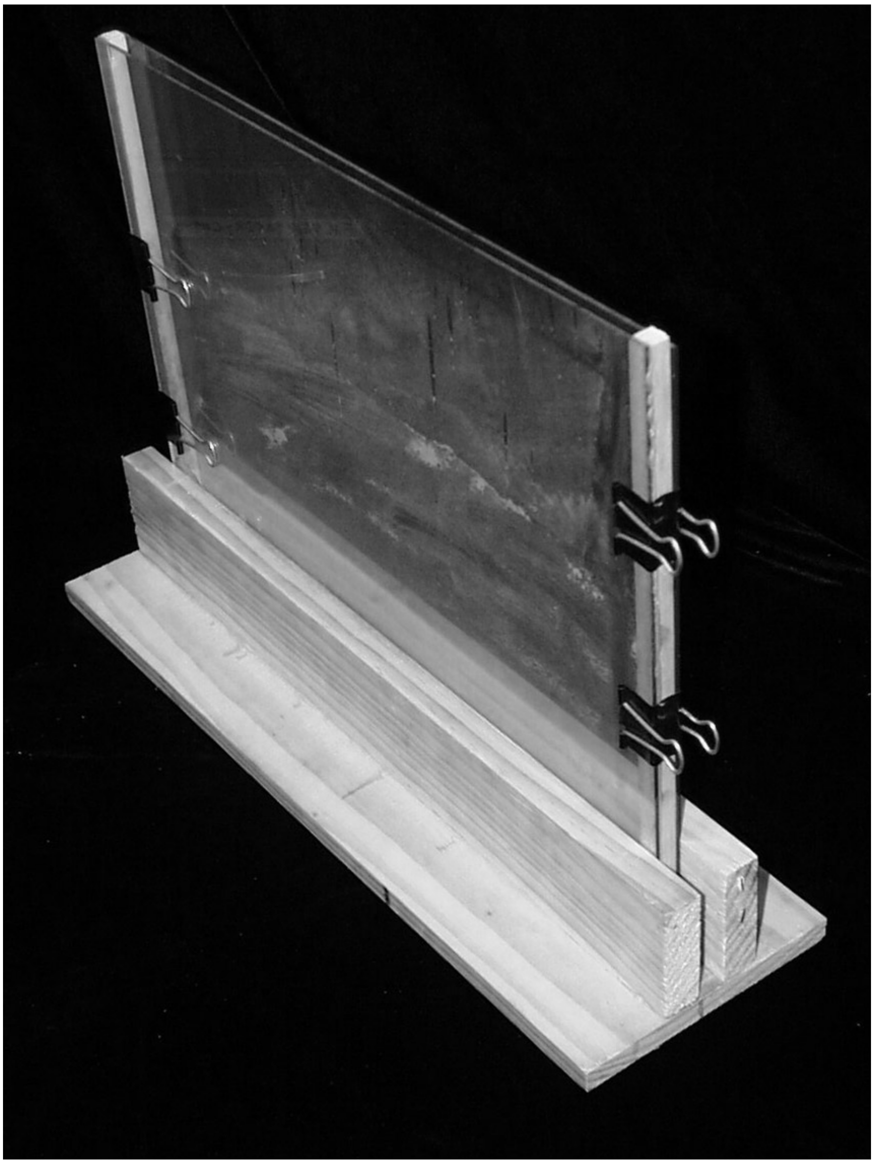


Figure 54. Glass-sided terrestrial observation cage held together with large binder clips.

This observation cage is economical and easy to construct. To make this cage you will need two pieces of glass or plexiglas, scrap lumber and bulldog clips (spring steel). Cut two pieces of plexiglas to the desired dimensions of the completed cage. Cut thin strips of scrap lumber to match the dimensions of the plexiglas in length and approximately 1/2" square. Starting with the sides, carefully line up the edges of the plexiglas and the strips of wood. Use bulldog clips to hold the pieces together. Once the sides are in place repeat the process with the bottom wooden strip. Silicone caulking can be used to more tightly seal the cage. To make a thicker cage use thicker pieces of lumber and larger bulldog clips. Be careful when using large bulldog clips with glass. A top can be made out of another scrap of lumber if needed.

Put soil into the cage and add the organisms you wish to study. If you leave several inches between the top of the cage and the soil surface, many soil dwellers will not attempt to escape from the cage. To better mimic subterranean conditions keep the sides of the cage covered with cardboard unless you are actively making observations. Try to use red or subdued light during observation periods. Alternatively sheets of clear red acetate or gel can be affixed to the cage sides with tape of clips. A simple stand can be constructed out of lumber to hold the cage in an upright position.

ANT FARMS

Ants are fascinating insects. In almost every habitat they are one of the most numerous organisms. They are important predators and play a major role in soil dynamics. They are among the best animal architects. Their nests can be huge and incredibly complex. Ants are also easy to rear in captivity and provide a rich resource for classroom activities. There are several types of ant farms that can be easily constructed.

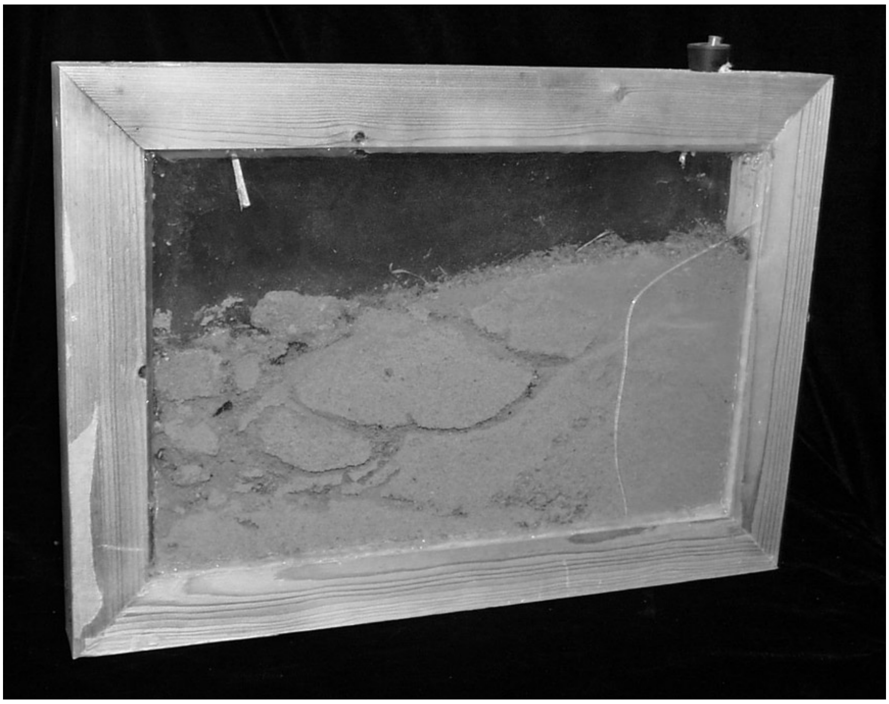


Figure 55. A simple ant farm, note the opening at the top for feeding the ants. A vinyl hose can be attached to allow the ants to forage outside of the farm.

Uncle Milton's ant farm has been sold for decades. The basic structure is a plastic frame with clear windows on both sides. A wooden ant farm with the same basic structure can be built from 2" X 2" lumber and two pieces of glass. Plastic can also be used for this project but glass is preferred. The activity of the ants and the abrasive quality of soil will quickly scratch plastic. First you must decide the size of the farm you desire and purchase two pieces of glass. Prior to cutting the 2" X 2"'s use a table saw to kerf (notch) the lumber along its length about 1/8" inside both edges on one side of each piece. The kerf should be about 1/4" deep and, if a standard saw blade is used, should be 1/8" wide, the exact thickness of standard glass. Cut the lumber to length and miter the corners to a 45° angle. Be careful when calculating the lengths of the sides. Remember the glass will fit into the kerf so the depth of the kerf must be taken into account during measurements. Assemble the bottom and sides with the glass in place. Make sure the corners are tight. Before attaching the top fill the farm about 3/4ths full with soil or sand. The space is left so the ants will have somewhere to put soil excavated from their tunnels. Use a large diameter drill bit to drill several holes along the top of the farm. These are used to add food and water. You can also attach hoses to some of the holes to provide a path in and out of the farm. A hose can lead to feeding containers or observation arenas made from a plastic shoebox or jar. When you are not actively observing the ants cover the sides of the farm with cardboard or clear red plastic. Ants will be more likely to tunnel near the glass if it is kept dark.

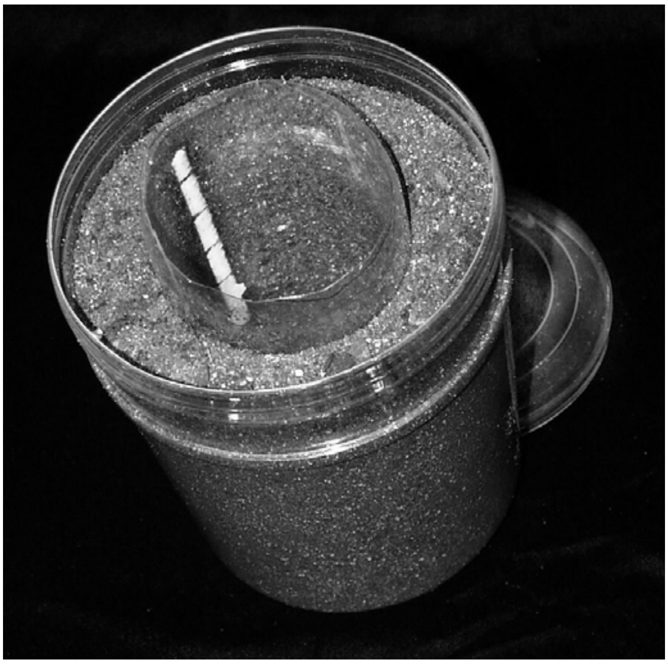


Figure 56. An ant farm constructed from a soda bottle and another larger container.

The soda bottle forces the ants to excavate around the perimeter of the larger container where they can be more easily seen.

An even simpler ant farm can be made from a two-liter soda bottle and another larger clear-sided container. Hold the two-liter bottle centered inside the larger container and fill in soil around the bottle until the container is full. If the large container doesn't have a lid smear a mix of petroleum jelly and mineral oil (1:1) around the rim to prevent ants from escaping. The inner bottle forces the ants to tunnel near the sides of the container where they can be more readily observed. A clear plastic shoebox can also be modified in a similar fashion if you cannot find a suitable cylindrical container.

For small ants an ant farm can be made from plaster of Paris or casting plaster. On a flat board lay out a series of trails and small chambers with modeling clay, use scrap lumber to build a frame (about 1 1/2" high) around the trails. Pour plaster into the frame and allow it to dry. Turn the cast over and remove the clay. Cut a piece of clear plastic to cover the top of the farm. Use petroleum jelly or clay as a sealant.

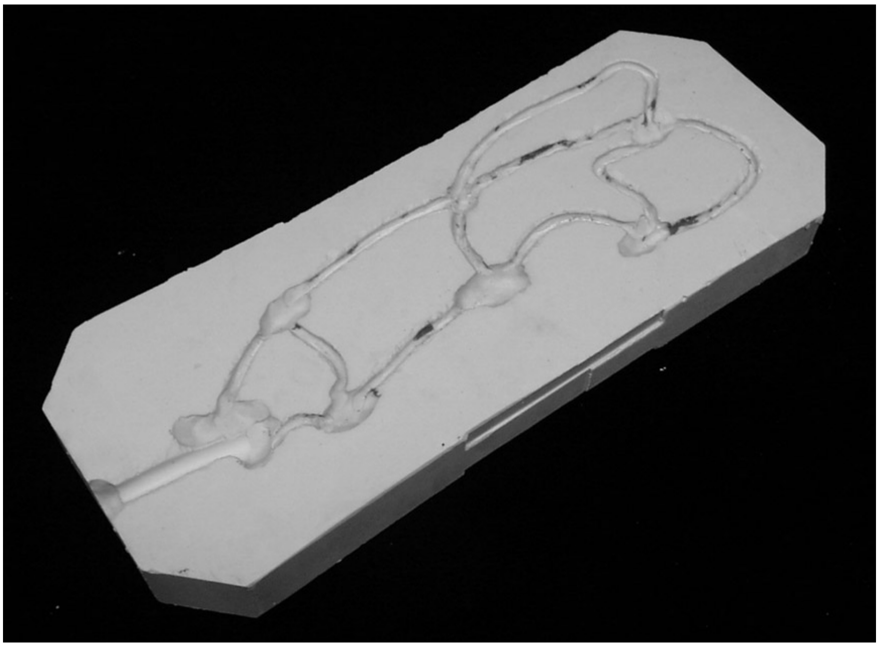


Figure 57. An ant farm made from plaster. A piece of glass or plastic is used to cover the galleries when ants are in residence but has been left out of this photograph for clarity.

HANDLING INSECTS AND PREPARING AN INSECT COLLECTION

If you undertake research projects such as those outlined in this book, it is a good idea to keep voucher specimens and develop a reference collection to facilitate identification of specimens. Even if you don't conduct a research project you may want to start collecting insects as a hobby. Insect collecting is popular worldwide and it is a great way to develop an understanding and appreciation of the diversity of life and the intricacies of ecosystems. Preparing an insect collection is fairly easy but there are some special techniques and procedures that should be followed. In this unit I will explain some the basics of preparing a collection as well as describing specialized equipment that will help you handle insects you collect.

Killing Jars

When you are collecting insects in the field it is important to have a killing jar handy to efficiently dispatch specimens you want to retain. A simple, yet effective, killing jar can be made using ethyl acetate (finger nail polish remover) as poison. To make a killing jar you will first need to pick a suitable container. The container should be made of clear colorless glass, if you use plastic make sure it is resistant to solvents or the ethyl acetate will melt it. In general you do not need a very large jar but a good jar will have straight sides with little or not shoulder at the mouth. The straight sides make it easy to pour small dead insects out of the jar. It also helps if the jar is at least 5"-6" tall. This helps prevent insects from escaping. My favorite jars are made from relatively tall but small diameter olive and caper jars. You may want to make one large jar for bigger insects like butterflies and dragonflies that have delicate wings.

Once you have selected the proper container prepare a mixture of smooth, slightly watery, plaster of Paris. Pour a 1"-2" layer of plaster in the bottom of the jar. Once the plaster has dried wrap heavy tape such as duct tape around the base portion of the jar just high enough to hide the plaster from view. Killing jars are bottom heavy and tend to break when dropped the tape will hold the pieces together if an accident occurs. "Charge" the killing jar with a couple of tablespoons of poison. The liquid will absorb into the plaster. Put a sheet of tissue paper into the jar and it is ready to go. The tissue serves a few purposes. It helps to minimize damage to fragile parts such as antennae by minimizing movement of insects in the jar. The tissue also absorbs body fluids produced by insects placed in the jar. Grasshopper "tobacco", for example, can be extremely messy. You should also clearly mark the sides and lid of your jar with the word poison just to be safe. One problem with this type of killing jar is that it rapidly loses its toxicity because the poison is volatile and readily escapes from the jar especially in hot weather. If you plan on a long collecting trip, it is a good idea to bring additional ethyl acetate along so you can "recharge" your jar in the field.

A more lethal and longer lasting killing jar can be made using hydrogen or potassium cyanide. Both of these can be purchased from chemical supply stores. They both produce highly lethal and fast acting cyanide gas. The advantage of this type of jar is that it remains efficacious for years and because of the rapidity of action reduces the possibility of an insect damaging itself before it dies. Many millipedes produce cyanide as a form of defense, old-time entomologists would actually collect millipedes at the start of a collecting trip and put them in a jar to make a "natural" killing jar. When preparing a cyanide killing jar it is important to work in a well-ventilated place, outside or under a fume hood are best.

Put about a 1/2” layer of cyanide in the bottom of your jar. On top of the cyanide put a 1” thick layer of wood shavings. Cover the entire surface of the shavings with a 1/2” thick layer of plaster. After the plaster is dry put the lid on the jar tightly, it is imperative to tape the end of the jar with several layers of heavy tape to minimize the chance that cyanide powder will be spilled if the jar is accidentally broken. With time the effectiveness of the jar may diminish. When that occurs add a small amount of water to the jar.

It is best to remove insects from a killing jar as they are collected. One good way to carry them is in a small Tupperware or Rubbermaid container. A sandwich-sized container is perfect. Before going on your collection trip fill the container with layers of tissue paper or paper towels. Try to find a brand that doesn’t have a lot of lint; kimwipes work great. Carefully remove the contents of your jar and, starting at the bottom of your container, spread them out on a layer of tissue. Be sure to include a slip of paper with collection data with each layer. Fold the tissue over. Each time you empty your jar put the specimens on a new layer of tissue. Keep the container full of tissue layers to minimize jostling your samples.

Pinning and mounting collected specimens

Not all insects that you collect are preserved in the same way. What you do with the insect after it is collected depends on the size, developmental stage and species. Immature stages are usually kept in 50-70% alcohol (ethanol, methanol and isopropyl will all work). Alcohol will make the specimens somewhat brittle so don’t use a concentration higher than 70%. Alcohol does not penetrate specimens very quickly so large larvae will often turn black due to bacterial and enzymatic activity that continues inside the dead insect. This can be prevented by using a penetrating fixative such as KAAD or formaldehyde (formalin and gluteraldehyde will also work). Always wear gloves and work in a well ventilated area when using these compounds. You can also prevent discoloration by boiling large larvae in water for several minutes, just long enough to shut down biochemical and bacterial activity inside the specimen.

Soft-bodied insects such as termites and aphids are also usually kept in alcohol. If soft-bodied insects are pinned, the abdomens will usually shrivel up making it very difficult to see morphological features. A good rule of thumb is that adult insects in Orders that have aquatic immature forms are placed in alcohol and not pinned (e.g. caddisflies, mayflies, stoneflies) but there are exceptions. If in doubt it is better to preserve the specimen in alcohol. It is a good idea to separate your sample into vials by species rather than keep an entire mixed sample in one large vial. Be sure to include collection data in each vial (more on that soon). There are several different types of vials that can be purchased for specimen storage. Rubber stoppered rimmed vials work well and can be used for long term storage with minimal loss of fluid but are difficult to open and close. Screw capped vials are easier to open and close but are not archival i.e. the alcohol will evaporate from the vials with time. Polyseal screw cap vials offer easy access to specimens and archival storage of specimens. Most general entomology textbooks will have extensive information on proper preservation techniques for each taxonomic group.

Pinning insects takes a steady hand and a little practice. Hardbodied adult insects are pinned. The simplest form of pinning is simply impaling the specimen with a pin. By convention insects are pinned through the right side of the body between the 2nd and 3rd pair of legs (meso- and metathoracic legs). The pinned specimen should be level on the pin front-to-back and side-to-side. The top of the specimen should be about 3/8” from the top of the pin providing a space to hold the pin without touching the insect (it is very fragile). A 3/8” piece of small diameter plastic or brass tubing such as a pen refill can be used as a simple pinning guide. When pinning a specimen push the pin through the insect leaving less than 3/8” of the pin projecting from the top. Take the guide and push it down over the head of the pin. With your finger over the end of the guide push gently down until the pinhead touches your finger. At that point the top of your specimen is 3/8” from the head of the pin. Insect pins come in different sizes; number 2 is the most commonly used. Thinner pins (e.g. 1, 0, 000) are used for smaller and thinner insects. The pin should never be larger than 1/4 the body width. Thinner pins will bend more easily when being pushed into a pinning surface such as an insect box. To minimize breakage try to pin your specimens when they are fresh. Insects become very brittle and fragile within days after being collected.

If your specimens are too dry for pinning you can “relax” them using the following method. Put about one inch of sand in the bottom of an airtight container that is large enough to hold a dinner plate. Soak the sand with water that has a small amount of vinegar added (a few tablespoons/quart). Rest the plate on top of the sand and put your specimens on a paper towel atop the plate. Close the container tightly. The specimens should be relaxed enough for pinning within a few days (depends on initial dryness and specimen size). Make sure that the specimens do not come directly in contact with the wet sand.

Minuten pins and paper points are mounting methods used for smaller specimens (see photograph below). Paper points are small triangular pieces of cardstock (sometimes clear acetate is used) made with a point punch, which is basically a specialized hole punch. The points are mounted on standard insect pins (again 3/8” from the top). To mount an insect on a paper point, bend a small part of the paper point tip down using a pair of fine forceps. Put a small amount of clear nail polish on the paper point and glue the insect to the point on its right side between the 2nd and 3rd legs. Be careful not to use too much glue and try to keep the point on the right side of the body. Your goal is to leave at least half of the insect unobscured so important structures can easily be seen. It may be helpful to put a slight curve in the bent down point tip so it cradles the insect’s body. A good dissecting microscope makes this entire process much easier.

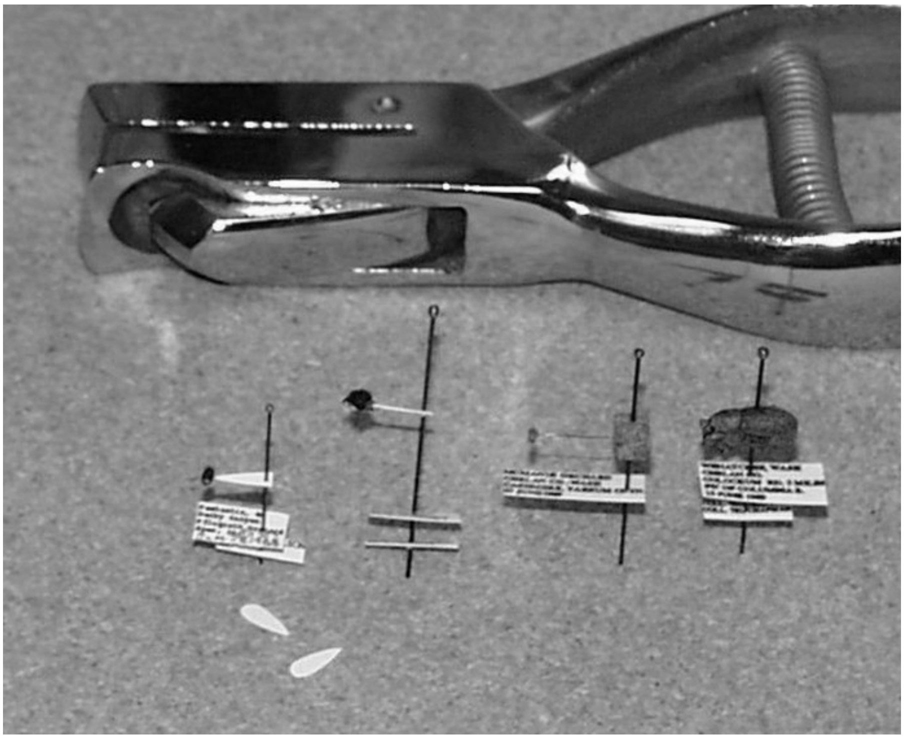


Figure 58. Paper point punch and specimens mounted (left to right) on paper points, minuten pins and standard pins, note the size and placement of the labels.

Minuten pins are another method for mounting small insects. Minutens are very sharp and of small diameter. The minuten is partially pushed into a block of cork or dense foam that is affixed to an insect pin (Figure 58, 2nd specimen from right). Lay the specimen on its left side (right side up) and pierce it through the right side with the minuten. Again it really helps to do this with the aid of a dissecting microscope.

When pinning or mounting specimens it is important to consider how the finished specimens will be displayed or used. If the specimens are intended for a research collection it is best to push the legs, antennae, etc. in close to the sides of the body to save space in storage boxes and minimize breakage. For display purposes you may wish to pull the legs and antennae out from the body so they can be better seen. You may also want to spread the wings of your specimens. You will need a **spreading board** to accomplish that task. There are several types of commercially available spreading boards including very economical styrofoam models (see Figure 59). Some companies will sell kits that allow you to assemble your own spreading boards from the parts and hardware they provide (below, center, note adjustable center slot). It is also fairly simple to make your own spreading board.

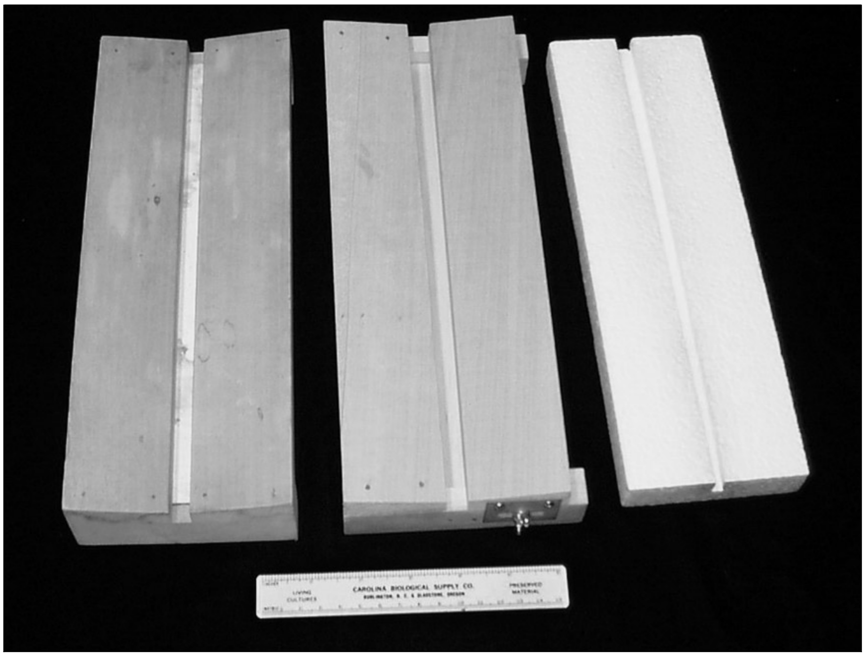


Figure 59. Spreading boards (left to right) homemade wooden, adjustable wooden made from commercially available kit, and commercially available Styrofoam board.

Balsa wood is an excellent choice of material for making a spreading board but you can also make a functional board from corrugated cardboard. The center slot of the board should be between 1/4” to 3/8” wide so it can accommodate the thorax and abdomen of most insects that typically have wings spread in collected specimens. Some kits have an adjustable slot area. The center rail of the board should be constructed from balsawood or another material that readily accepts pins (e.g. cork) and be mounted about 3/4” below the top of the wing spreading surface. The spreading surface can be flat or slightly beveled to give the wings a slight upward attitude in a completed specimen.

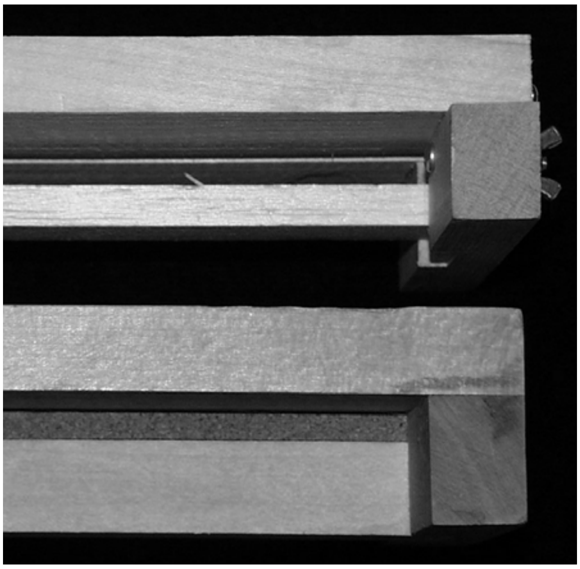


Figure 60. Side views of spreading board with balsa wood center rail (top) and basswood center rail topped with cork (bottom).

Generally only the wings of Lepidoptera (butterflies and moths) are spread but the wings of other groups of insects can also be spread to create interesting and educational displays. Spreading the wings of a beetle or bug can be tedious but the end results can be fascinating. Spreading the wings of any insects takes patience and practice. Most general entomology textbooks will provide instructions in the proper techniques to use.

If you want to spread the wings of very small insects use **mini-spreading boards** made from small blocks of lumber about 1 1/2” on all sides, the size of a child’s block (see Figure 61). Use a power saw to cut a notch (kerf) about 1/2” deep completely across the center on one side. It is safer to notch one long piece of lumber and cut it into smaller pieces after wards. If you are using a standard saw blade the kerf will be 1/8” wide. Drill a 1/16” diameter hole completely through the block down through the center of the kerf to accommodate insect pins. Pack the hole with soft clay to hold the pins securely in place. On each side of the kerf make 2 or 3 small notches in the edge of the block with a razor blade or knife. To complete the mini spreading board you will also need a length of fine thread long enough to go completely around the block 3-4 times. To use the block, place a pinned specimen into the kerf. Spread the wings carefully using insect pins. As each wing is moved into place secure it gently with the thread. The notches will hold the thread in place on the block.

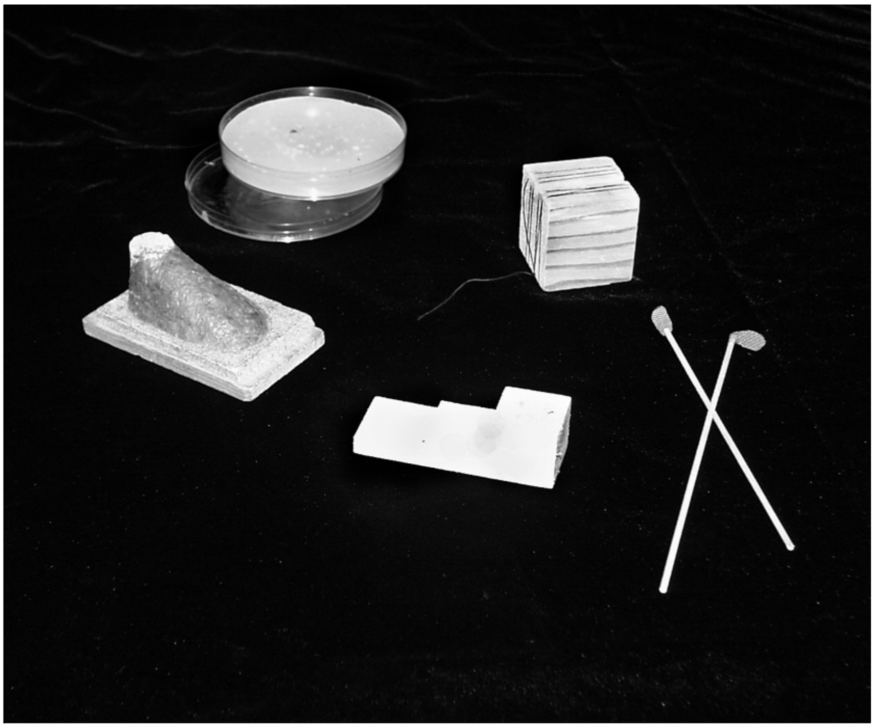


Figure 61. Useful devices for working with insects (clockwise from right); tools for removing small organisms from vials, pinning block, observation block, mini-dissecting tray and mini-spreading board.

There are several **other useful tools** shown in Figure 61. In the lower left are sticks modified into a device to facilitate removal of small specimens from vials of alcohol. The device is simply a tear-drop shaped piece of wire or nylon screen glued to the end of a small dowel. Small specimens can be easily lifted out of a vial without the need to pour out the vial or use a pipette. At the bottom center of the photograph is a **pinning block**. It is used to facilitate uniform labeling of pinned insect specimens. Each step of the block has a specific purpose, by convention the steps are 1/2", 3/4", & 1 1/8" with a hole, large enough to accommodate an insect pin, drilled completely through the block in the center of each step. The highest step is used to place insects at the correct height on a pin and is an alternative for the technique I described previously using a small piece of tubing. The middle step is the proper height for the primary label and the bottom step sets the height for a secondary label. To use the block, place it on a hard flat surface and put a label on the middle step centered over the hole. Push a pin through the label until the pin hits the hard surface. The label is now 3/4" from the pin bottom.

In the lower left of Figure 61 is an **observation block** used to hold pinned specimens. This device is especially useful when you are examining specimens with the aid of a microscope. To make an observation block you need a #4 cork, beeswax (clay will work), a small piece (about 1 1/2" X 2 1/2") of 1/4" thick plywood and a piece of sheet cork (1/4" thick) about the same size as the plywood. Glue the sheet cork to the top of the plywood. Glue the #4 cork, wide end down, near one end of the sheet. You can also drive a 1" finishing nail into the cork from the underside of the plywood if you want an extra secure connection. Form a mound out of beeswax (warm it up in your hands) or modeling clay that embraces the #4 cork. The observation block allows you to easily adjust and hold a pinned specimen in a steady position at almost any angle.

The final device shown in Figure 61 is a **mini dissection tray**. Similar in design to the wax bottomed metal trays commonly used during dissections of larger animals the mini tray is constructed from a standard disposable petri dish in which a thin layer of beeswax has been poured. The mini tray is small enough to easily fit on the stage of a dissecting scope. If the bottom becomes too rough from pinholes remelting the wax in a microwave renews the surface. Freshly killed crickets and cockroaches are fascinating dissections. With the aid of a dissecting microscope students can see Malpighian tubules and minute branches of the tracheal system. If the dissected insect is submerged in a weak saline solution (Ringer's) the movement of the Malpighian tubules can be observed. Make a wet mount (use saline) of the seminal receptacle of a female. Slightly mash and macerate the organ before placement of the cover slip. Under high power it is possible to see living, moving cricket sperm.

When collecting insects it is important to keep good collection data. The data you collect is reflected on the **insect label**. There can be several different labels on each specimen. The primary label usually contains the following information: date of collection, general location information, specific location information, habitat information, and collector's name. Here is a sample label:

20 October 2003
South Mountain, PA: Franklin Co.
In Rotten log nr. Walker cr.
Coll: G. S. Paulson

Additional labels can be added to include additional locality information or a species determination. Sometimes the collector's name is placed on a separate label. Generally there should be no more than 5 lines/label. If a specimen is paper pointed or on a minuten, the pin should go through the label near one end of the label rather than in the center. Examine the photograph of pinned specimens on page 103. Labels should be as small as possible but still legible and trimmed as close to the writing as possible to minimize their size. Large labels waste storage space and also can cause a lot of damage to other specimens if you aren't careful. Labels should be printed with waterproof ink. Laser printers will also work. Use high rag paper of a heavier than standard weight. Mount the labels on the pins so they all face in one direction with the text running in the same direction as the long axis of the insect's body.

Store your collection in an airtight case. There are a large variety of types and sizes available that can be purchased to fit most needs and budgets. You can also make your own by gluing a cork or foam bottom into a plastic storage container. Glass topped cases are especially nice for classroom displays and demonstrations. For large collections insect drawers are the most common method of storage. Be aware that insect drawers usually come in two standard sizes, U. S. National Museum and Cornell. Insect drawers usually have a glass top. Insects can be pinned directly into the bottom of a drawer or may be pinned into unit trays. Unit trays are small boxes with foam bottoms that come in several different sizes that can be used in a myriad of combinations but will exactly fit the inside of a drawer. They add a lot of utility to a drawer system by providing easily removable subdivisions of each drawer. Through the use of unit trays you can have as many as 32 subunits of each drawer.

No matter what type of case you use it is very important that it seal tightly to protect your specimens from infestations of insects and also from humidity. It is a good idea to place moth flakes or balls inside of each case to help prevent infestations. Make sure to secure the material so it doesn't accidentally damage specimens.

Another excellent display technique is a **Riker mount**. This is a shallow glass-topped case with a thick layer of cotton batting inside (quilting material will work). Insects are not pinned in a Riker mount but are held in place by the cotton. Riker mounts are commonly used to display specimens with spread wings or to illustrate life histories. Riker mounts can be purchased in many sizes ranging from the size of a paper backed book to almost 24" square.

A nice Riker mount can be made from a stationary box, a piece of glass (or plexiglas) cut to exactly fit the inside of the box lid, and a piece of batting cut to exactly fit the inside of the box bottom. Remove most of the top of the lid, leave a lip about 1/4" –1/2" wide completely around the lid to hold the glass (Figure 63). Paint the box a color of your choosing. Black is the standard color. Run a bead of silicone sealant around the lip on the inside of the lid and carefully lay the glass in place. While the sealant is drying spread the batting on the bottom of the box, there should be enough so the glass slightly compresses it when the lid is put on the box. You can put moth flakes under the cotton to protect against infestations. Carefully place your specimens on the batting. It will be difficult to make any adjustments to the initial placement. Carefully put the lid in place seal the mount shut with tape.

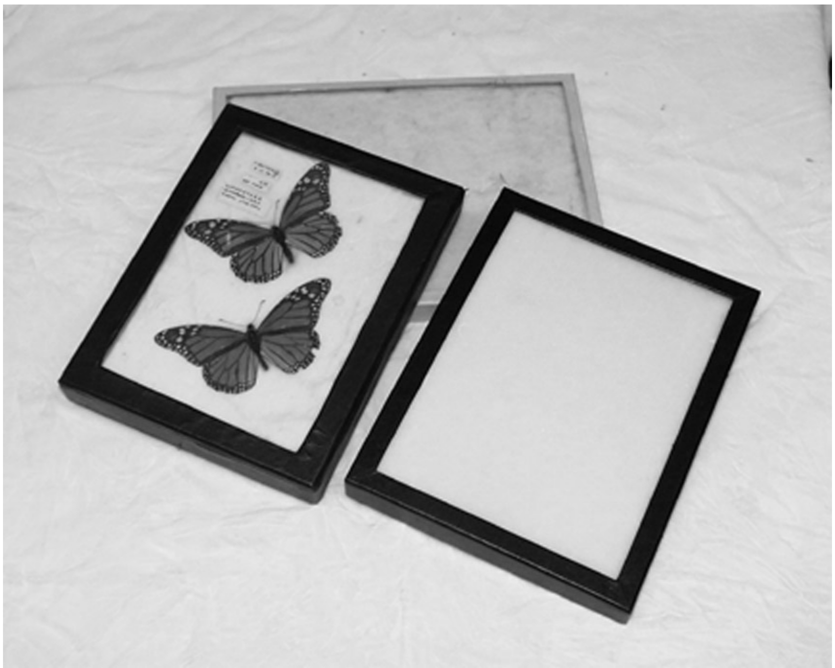


Figure 62. Riker mounts, those at top center and right are home made.

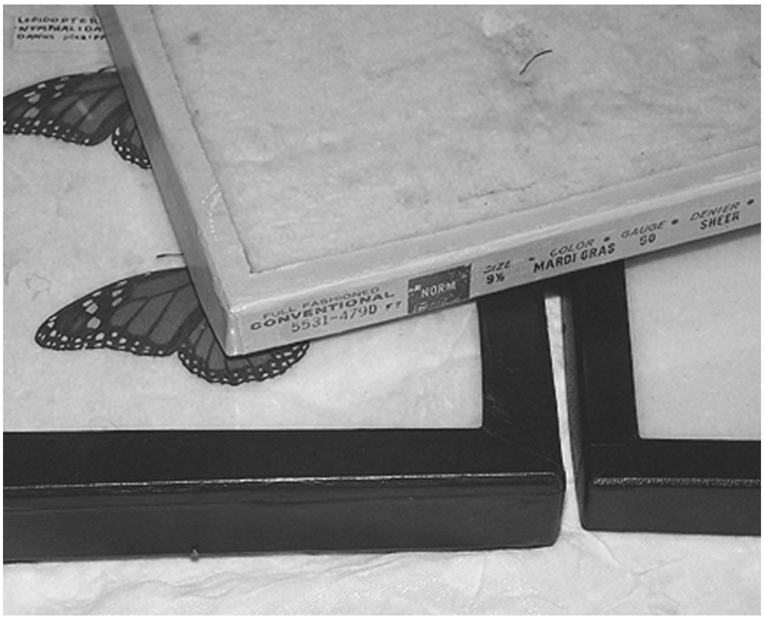

FULL FASHIONED
CONVENTIONAL
5531-4790
NORM
SIZE
9½
COLOR
MARDI GRAS
GAUGE
50
DENIER
SHEER

Figure 63. Detail of homemade Riker mount (top) and commercially produced mounts.

Magnifiers

Magnifiers or hand lenses are essential tools of the trade for entomologist. The best quality magnifiers can be very expensive but economical alternatives

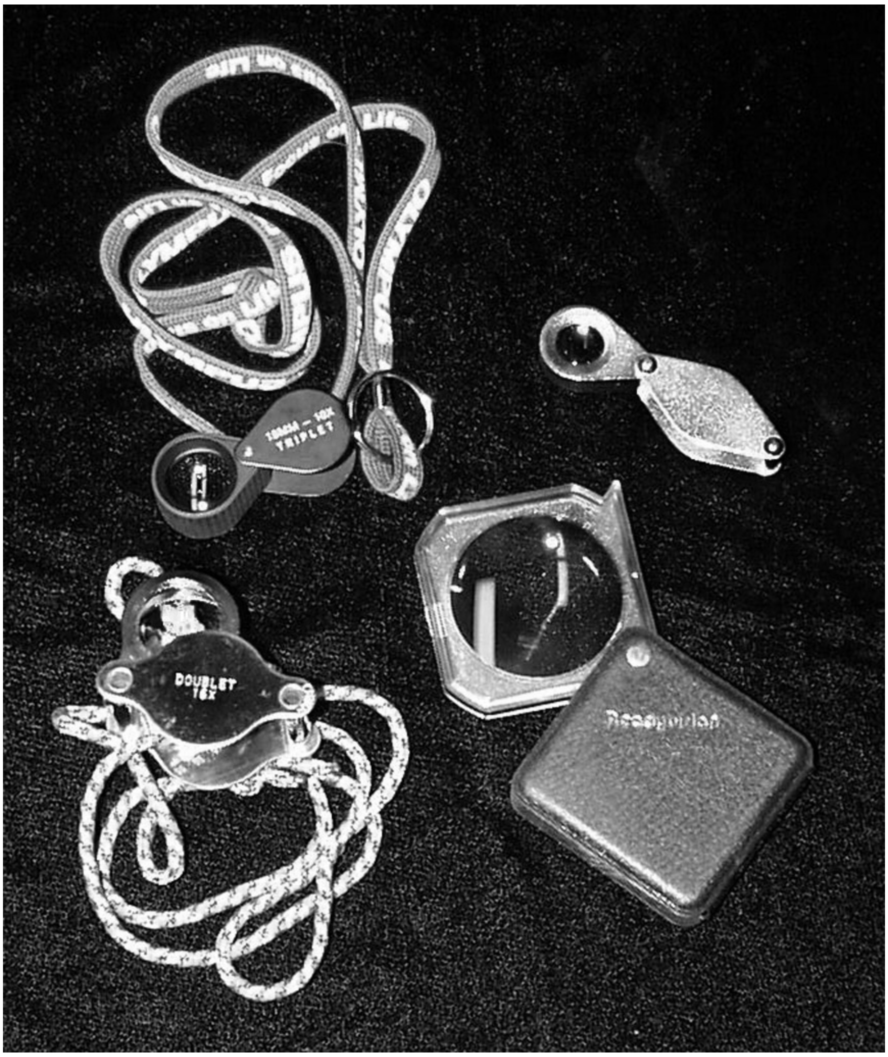


Figure 64. Types of magnifiers (clockwise from lower right); single plastic lens, doublet magnifier, 10X triplet magnifier and 15X triplet magnifier.

are available. The cheapest are made from a single plastic lens. These are durable but the plastic lens will scratch fairly easily and there is a lot of spherical aberration (distortion) especially near the edges of the lens. Doublet magnifiers have two lenses, which reduces distortion, but are quite a bit more expensive than a single lens. The best magnifiers are triplets, which reduce distortion to a negligible level by using three lenses. A general rule of thumb is that the higher the magnification the smaller the diameter of the lens (compare the size of the two triplet lenses in Figure 64, left 10X, right 15X). For most field purposes a 10X magnifier will suffice. It is a good idea to attach your magnifier to a lanyard or string and wear it around your neck; this keeps it easily accessible and reduces the likelihood of losing it.

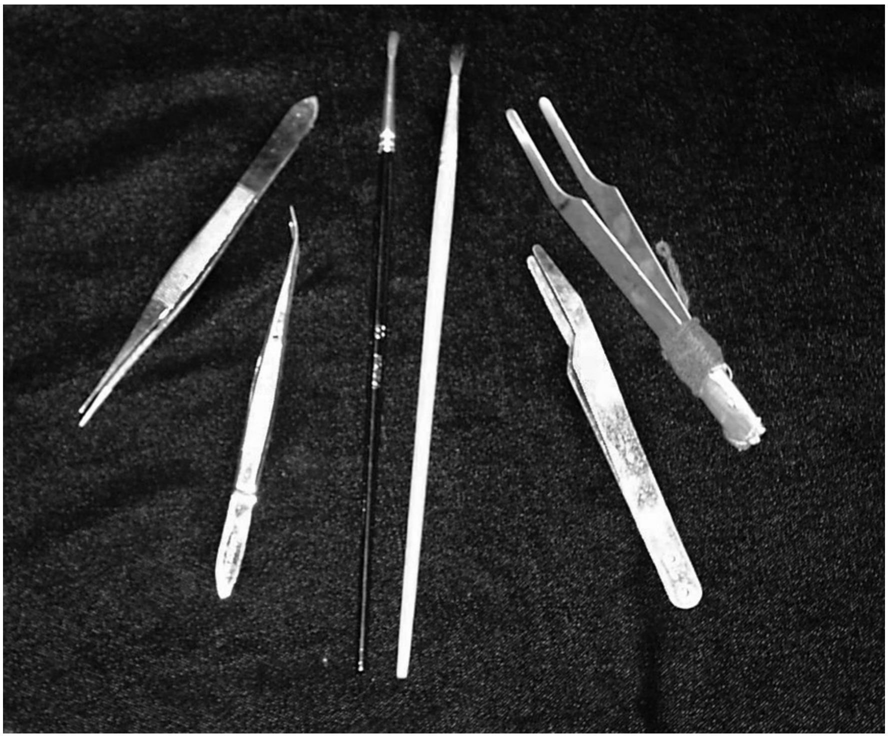


Figure 65. "Tools of the trade" (from left) forceps, fine point paint brushes and "soft touch" forceps.

Forceps and Paint Brushes

Handling insects without damaging them or you can be difficult without the correct tools. In the field and laboratory the most useful tool is a pair of fine point forceps with either straight or curved tips. Feather touch or larval forceps (Figure 65, both pairs on right side) are made of flexible metal that makes it virtually impossible to damage a specimen while handling it. These are especially nice for handling soft -bodied insects such as caterpillars or other immature stages. I attach my forceps to a long piece of string and wear them around my neck when in the field to keep them readily available and to prevent me from losing them. Paintbrushes are great tools for handling small insects. Number 1 or smaller (0, 00, 000) brushes are the best. To pick up an insect, moisten the brush on your tongue and gently touch the bristle to the back of specimen. A gentle tap on the brush is usually enough to dislodge the insect.

Tissue culture and ELISA plates are great for sorting specimens. They are economical and available from many vendors. They can be purchased with 6-96 wells. The smaller wells are not as useful as the larger.

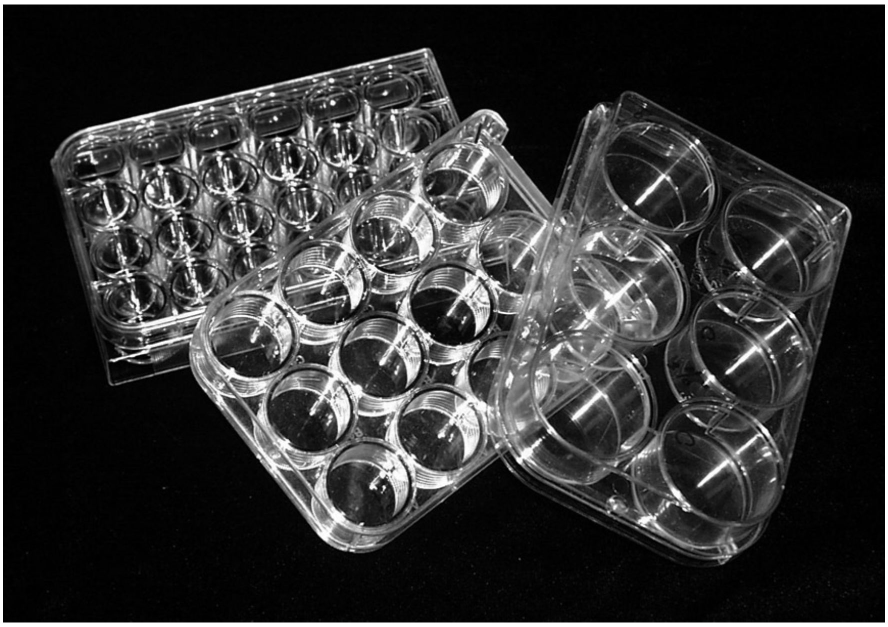
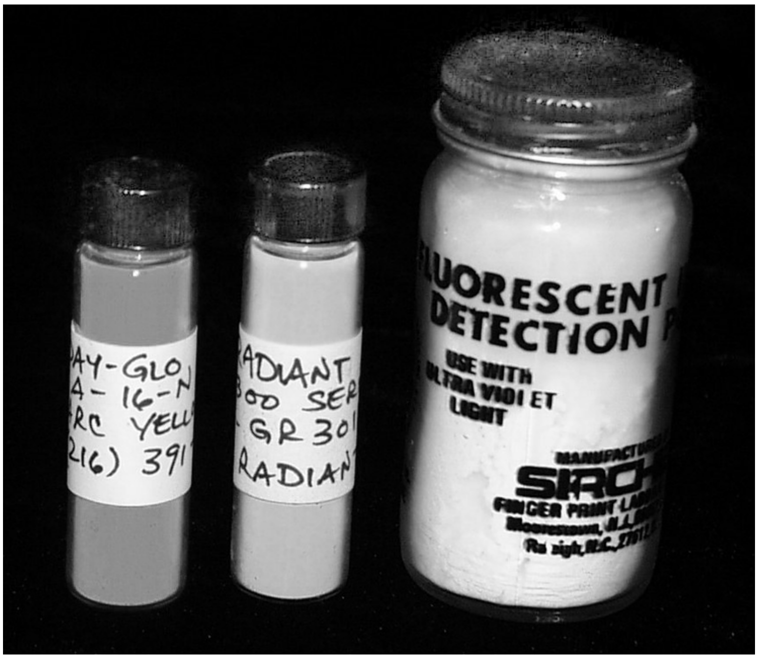


Figure 66. Tissue culture plates are very useful for sorting specimens.

CHAPTER 5

MISCELLANEOUS TECHNIQUES


AY-GLO
4-16-N
RC YELL
216) 391
ADIANT
00 SER
GR 301
RADIAN
LUORESCENT
DETECTION
USE WITH
ULTRA VIOLET
LIGHT
SIRCH
FINGER PRINT LA
Moorestown, N.J.
Raleigh, N.C.

MARKING ORGANISMS

Marking insects will allow you to study their dispersion and movements in a habitat as well as provide a basis for estimating population density using the Lincoln Index (see Appendix B). Before carrying out a study it is important to test the marking technique in a laboratory setting to make sure that the mark is not ambiguous, has enough permanence for the needs of your experiment and that the mark does not contribute to mortality of individuals.

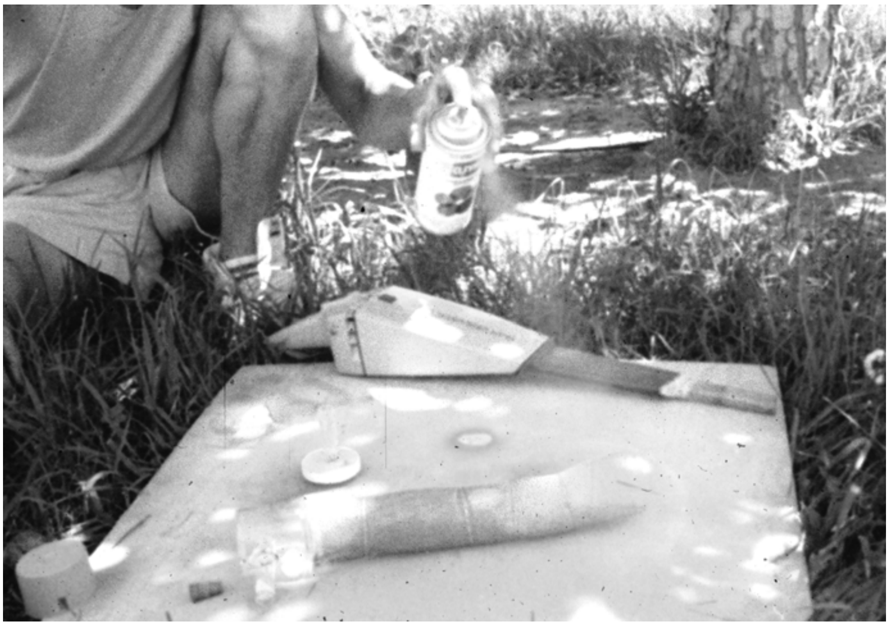


Figure 67. One method for marking a group of insects.

Insects can be marked in a number of ways and the simplest techniques use paints and colored powders. Paint can be applied to individuals with a brush or a group of individuals can be marked at once using spray paint (see Figure 67). Groups of insects can be released in the center of a piece of plywood and marked as they move to the edge of the board. By spraying the paint from several feet away individuals will be hit by only a few small droplets of paint. The protocol that I use includes a time limit for the insects to move off of the board. The assumption was that any insects remaining at the end of the designated time interval were not healthy enough to be included in my marked group. I always used fluorescent paint because a black light could then be used to facilitate locating marked individuals during recollections. If you decide to mark individuals, a vacuum device (Figure 68) can be used to hold larger insects in place during marking. The individual to be marked rests on the screened area. A vacuum source is attached to one pipe while the other is the air intake. The suction through the screen is sufficient to hold even the largest insect firmly in place during marking.

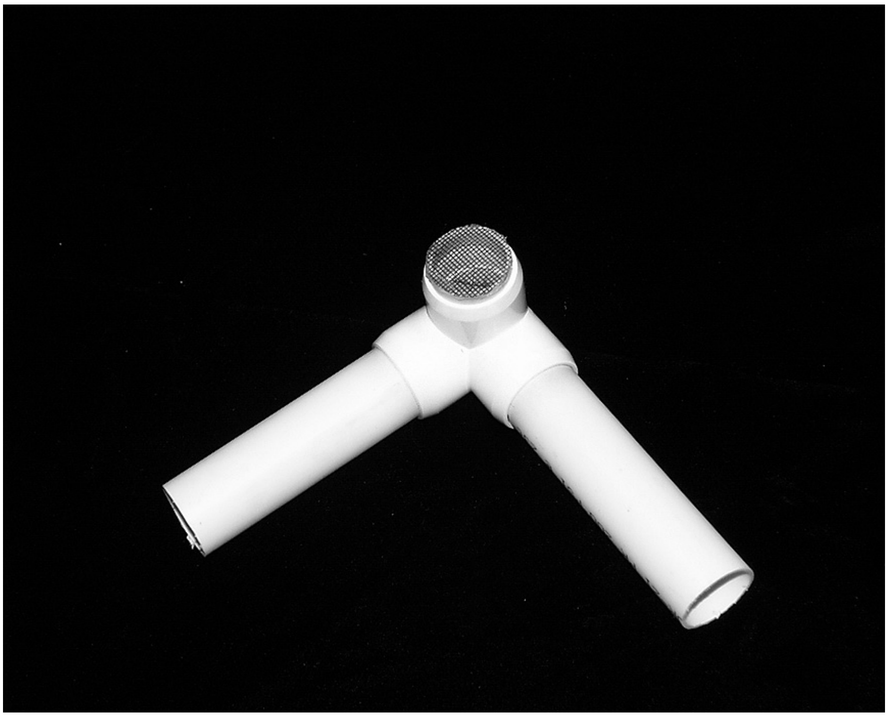


Figure 68. A device for holding large insects while marking, a vacuum is applied to one end of the device holding the insect in place on the screened area (upper center).

Powders are also a good way to mark insects but work best with insects that have abundant hair and/or scales such as flies, bees, and mosquitoes. Powdered tempra or poster paint works very well and is economical and easy to purchase. Fingerprint powders are finer so they tend to be more persistent and are fluorescent in black light facilitating locating marked individuals. Powders can be used with an emergence trap (next unit) to easily mark adult insects.

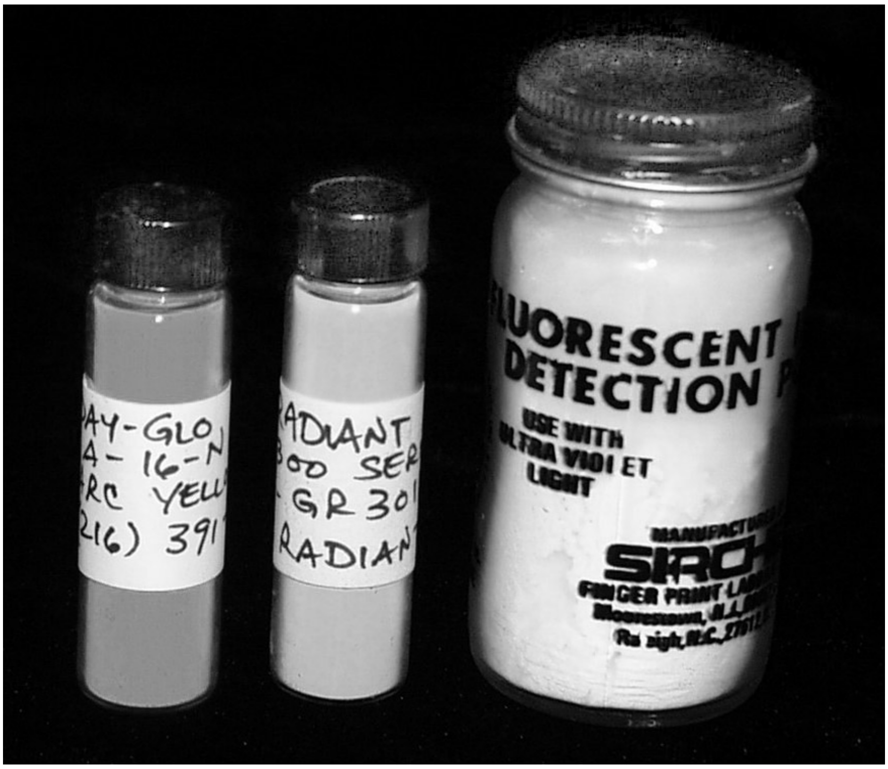

AY-GLO
4-16-N
RC YELL
216) 391
RADIANT
800 SER
GR 301
RADIANT
LUORESCENT
DETECTION P
USE WITH
ULTRA VIOLET
LIGHT
SIRCH
FINGER PRINT LAB

Figure 69. Tempra paint and finger print powders used to mark organisms. Fluorescent colors are easier to detect.

EMERGENCE TRAPS

An emergence trap can be used to collect insects from galls, pupae, parasitized material, etc. or as part of a marking technique. Emergence traps can be made from a variety of container types but I think the easiest method is to use two jars of identical size. A nice emergence trap can also be made from 2 liter soda bottles however. The trap Figure 70 is made from 2 – 10 ounce plastic jars (approximate 3” diameter x 4” high) with metal screw caps. To make a trap you will also need a funnel of appropriate size for your containers. Cut a hole in the center of both jar lids to accommodate your funnel. I used a plastic funnel in the trap pictured here and cut the end off of the funnel to leave a larger opening and so the funnel did not extend too far into the upper jar. The funnel projects into the upper jar about 1”. Glue the jar lids together and glue the funnel into the opening. Seal both sides with silicone. The completed trap is two jars with an inverted funnel between them.

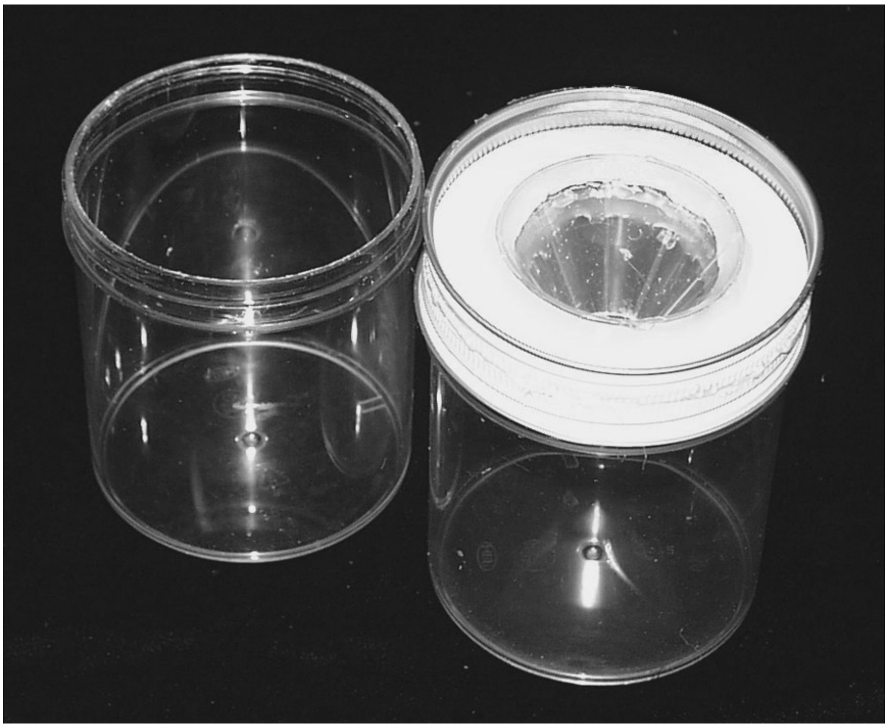


Figure 70. An emergence trap constructed from two jars, the jar on the left fits on top of the other jar when in use.

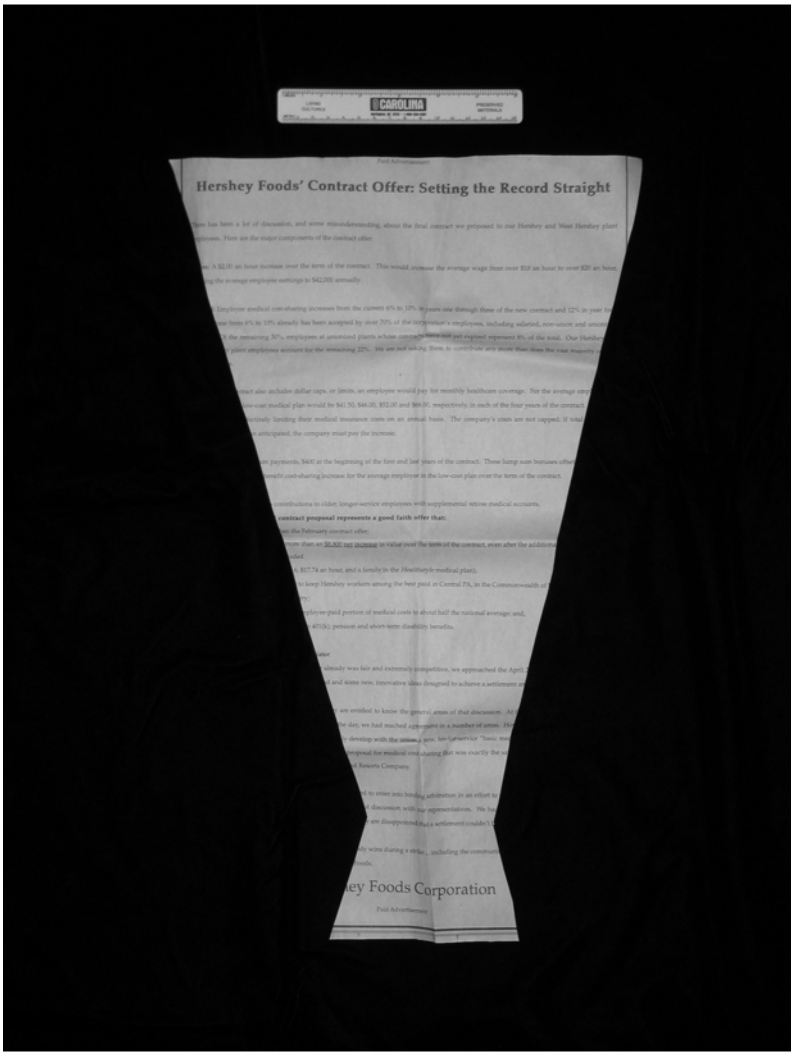
Traps of almost any size can be made to allow larger samples to be processed. The top container does not have to be the same size as the bottom container since it is only holding insects that have flown out of the sample. For example a 10 ounce jar such as shown in the illustration could be affixed to a 1gallon jar or even a 5-gallon bucket if larger amounts of material are being sampled. The top container also does not need to be constructed of the same material as the bottom container. In fact efficacy of the trap may be increased if the bottom container is opaque and the top container is clear, as insects will be attracted to the collection chamber light source.

An emergence trap is very useful for collecting small insects such as parasitic wasps and gall making flies. Place the material of interest into the bottom container and assemble the trap. As organisms emerge they will fly up into the upper jar and become entrapped. Almost anything can be placed into the emergence trap even aquatic samples. Aphid or scale infested leaves and sticks will often yield several species of parasitic insects as well as the adult hosts. Galls often house a diversity of insects, as do eggs and pupae of other arthropods. Emergence traps help reveal the hidden diversity of an ecosystem and the complexity of a "simple" niche like a gall. Each student can provide their own samples that can be monitored in class each day.

An emergence trap can also be used in conjunction with marking powders for quick and efficient marking of adult insects. To use a trap in this manner you will need thin strips of paper such as those produced by a shredder. Place some of the shredded paper in a zip-lock plastic bag with a small amount of marking powder. Shake the bag thoroughly until the paper is covered with powder. Put the colored paper into the upper chamber of the trap until it is about half full of loosely packed paper. As insects emerge from the lower chamber they will pick up the marking powder as they move up through the paper. This technique works extremely well with small moths, flies and mosquitoes and can be used for mark-release-recapture studies (MRR). See Appendix B for an explanation of MRR and some research ideas.

APPENDIX A: NET PATTERN

Plankton net pattern: You will need two identical pieces of net to produce a net. A collection funnel attaches to the narrow end. Larger (or smaller) nets can be produced by proportionally changing the size of the pattern. The ruler in the photograph is approximately 6” (15 cm) long.


CAROLINA
Hershey Foods' Contract Offer: Setting the Record Straight
ey Foods Corporation

APPENDIX B: STATISTICAL ANALYSES

Once the samples have been collected and processed what can you do with the resulting data? The tendency in classroom situations is to do simple comparisons through graphing or calculated means. The problem with such a simple approach is that it doesn't allow researchers to understand when differences in numerical values reflect actual (statistical) differences between experimental groups or treatments. In other words, a difference is not always a difference. The magnitude of difference between the values in question is not always a good indication of the relationship between experimental groups. Statistical analyses help take the guesswork out of interpreting data. Simple statistical analyses are not beyond the scope of most classrooms and they allow researchers to discuss their results in terms of probabilities that one group of data is different from another or that a hypothesis is supported or refuted. Before undertaking a project utilizing some of the apparatus or techniques described in this book a researcher should have a well thought out research plan including a hypothesis that is being tested and an idea of how the resulting data will be analyzed.

Two relatively simple and frequently used statistical analyses, Analysis of Variance (ANOVA) and Student's t-test, will probably cover the "statistical needs" of anyone using techniques presented in this book. The t-test is used to compare sample means of two sets of data. ANOVA, as the name implies, is used to examine the variance in sample data from two or more groups. There are several types of ANOVA that allow for very complex evaluation of data. The fairly simple one-way ANOVA will probably be most useful for teachers. Both of these analyses can be performed using the "Data Analysis" feature (found in the "Tools" menu) of Excel but there are many other statistical analyses programs that can be used.

On the following pages are samples of the analysis tables produced when conducting a t-test or ANOVA using Excel. As mentioned previously, statistical analyses provide probabilities (P or P-value) that differences in our data are real not simply due to sampling error or random variation in populations. By convention a probability of 0.05 or less is considered statistically "significant". This means that there is a low probability that an observed difference is due to chance alone

The following table is the product of an Excel t-test on the diameter of ant mounds from two different habitats, forest and meadow. The analysis indicated that nest diameters of these two groups were statistically different from each other. There are two P values listed on the table, one for a two-tailed test, the other for a one-tailed test. The term "tail" refers to the high and low value regions (in this case the largest and smallest nest diameters, respectively) of a value distribution (which is generally bell-shaped) curve. A two-tailed test is used if you are trying to determine if the average value for a particular characteristic (in this example diameter of ant nests) from one sample population differs from that of another sample population, not specifically larger or smaller, just different. The one-tailed test is used when you are hypothesizing that the average value obtained for one sample population is larger (or smaller) than that of the other sampled population.

t-Test: Two-Sample Assuming Equal Variances		
	Forest	*Meadow*
Mean	25.000	35.455
Variance	37.400	51.673
Observations	11.000	11.000
Pooled Variance	44.536	
Hypothesized Mean Difference	0.000	
df	20.000	
t Stat	-3.674	
P(T<=t) one-tail	0.001	
t Critical one-tail	1.725	
P(T<=t) two-tail	0.002	
t Critical two-tail	2.086	

On the following page is an example of a table produced using ANOVA to examine aphid populations in outer vs. inner foliage of a tree. Once again our P value indicates that there was a significant difference between aphid populations on inner vs. outer foliage.

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Outer Foliage	11	390	35.45	51.67
Inner Foliage	11	275	25	37.4

ANOVA

Source ofVariation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	601.14	1	601.14	13.50	0.001	4.35
Within Groups	890.73	20	44.54			
Total	1491.86	21				

Mark-Release-Recapture (MRR) studies can provide interesting information about the movements of organisms. The protocol is that organisms are caught and marked or tagged in some fashion before being released. During subsequent collections the mark allows the researcher to identify previously collected organisms and learn about its movements in the study area. MRR techniques can be coupled with simple mathematical analysis to provide information about the population size of the organism being studying. The Lincoln Index is commonly used. To use the Lincoln Index it is important that the exact number of organisms marked during the initial collection is known. Here is one version of the Lincoln Index:

$$N = \frac{T(n+1)}{t+1}$$

Where N = total population, T = the number marked and released, n = the total number of organisms in the recapture sample (marked and unmarked), and t = the number of marked organisms in the recapture sample. Using this formula an estimate of population size can be obtained.

The final methods of analyses that are applicable to the type of data collected with many of the techniques in this book are diversity indices. Most of these are simple comparisons of the number of species collected versus the number of individuals of any species collected. There are many different indices that are used by ecologist two of the most commonly used are the Shannon Index and the Simpson Index. The Simpson Index is probably the easiest to understand:

$$D_s = \frac{N(N-1)}{\sum n(n-1)}$$

Where species diversity is D_s, N = total number of individuals of all species, and n = number of individuals of one species ($\sum$ means sum, meaning you calculate the n(n-1) value for each species and add them all together). The index has a value of 1.0 for a community in which every individual is of one species and ranges to infinity in a community in which every individual is of a different species.

Using these few simple analyses, teachers can help students better understand the nature of science and ecosystems.

APPENDIX C: REFERENCES & FIELD GUIDES

There are an incredible number of books about insects that are available. I've selected three "desert island" books.

American Insects: A Handbook of the Insects of America North of Mexico, R. H. Arnett. This book is a wealth of knowledge; it includes a lot of illustrations, collection methods, taxonomic keys, etc. It can be very useful for identification below the Family level. I've always felt that the use of "Handbook" was deceiving. This is a huge book good for Jack Lalane type workouts when you're not looking something up.

Introduction to Insect Biology and Diversity, H. V. Daly, J. T. Doyen & A. H. Purcell. A good general reference with good keys as well as detailed information on biology, life history, etc.

An Introduction to the Study of Insects, D. J. Borror, C. A. Triplehorn & N . F. Johnson. An excellent general reference with keys and detailed information about biology, life history, etc. This book was the standard against which others were measured for many years. It may be out of print soon.

FIELD GUIDES

A Field Guide to the Insects, D. J. Borror and R. E. White. This is still my favorite guidebook it is concise and easy to use. No photographs just drawings.

Simon and Schusster's Guide to Insects, R. H. Arnett & R. L. Jacques. This book has nice photographs of insects but I find it difficult to use for identification.

The Audubon Society Field Guide to North American Insects and Spiders, L. Milne & M. Milne. Again, very nice photographs and a waterproof cover but I think it is difficult to use for identification purposes.

Dorling Kindersley Handbooks: Insects, Spiders and Other Terrestrial Arthropods, G. C. McGavin. Not really a field guide and of limited usefulness if you are interested in only North American species but this is still a great book that contains lots of good information and wonderful photographs.

William C. Brown Co. also has a large series of "How to Know" books that provide keys to various taxonomic groups from tapeworms to trees, these books can be very useful to a knowledgeable user but can be ponderous to a novice. They are also out of date so some of the taxonomy is incorrect. These are still very handy and since they are relatively cheap you can have a great reference library for a reasonable price.

APPENDIX D: SOURCES FOR MATERIALS

Aquatic Research Instruments: http://www.aquaticresearch.com
Water samplers, plankton nets, Surber samplers, Hess samplers, drift nets, calibrated lines, armored thermometers, BOD bottles.

Ben Meadows: http://www.benmeadows.com/
Waders, rubber boots, field water test equipment, kick nets, dip nets, wash buckets, forceps.

Bioquip: http://www.bioquip.com
Entomological equipment and books.

Carolina Biological Supply: http://www.carolina.com
General biological supply company. Flexible arm magnifiers, hand lenses, forceps, kick nets, microscopes, reagents,educational materials, live and mounted specimens for instruction.

Consolidated Plastics: https://www.consolidatedplastics.com/
Sampling trays, buckets, nalgene bottles, garbage bags, Whirl Paks ®.

Fisher Scientific: https://www1.fishersci.com/catalogs
Lab equipment, sample bottles, sieves, reagents, incubators, water test equipment, Whirl Paks ®.

Forestry Suppliers, Inc.: http://www.forestry-suppliers.com
Field equipment and supplies.

Gempler's: http://www.gemplers.com
Field equipment and supplies.

LaMotte: http://www.lamotte.com/
Water sampling kits, field and lab water testing equipment, Secchi disks, water samplers, armored thermometers, calibrated lines, plankton nets, kicknets, educational materials.

United States Plastic Corporation: www.usplastic.com
Every imaginable type of plastic, tubing, sheets, etc.

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